VOL. 36, PART 4 DECEMBER 1953

The British Mycological Society

(Recognosce notum, ignotum inspice)

TRANSACTIONS

Edited by

P. H. GREGORY and G. C. AINSWORTH

CAMBRIDGE UNIVERSITY PRESS
LONDON: BENTLEY HOUSE, N.W.1
NEW YORK: 32 EAST 57TH STREET, 22

OFFICERS AND COUNCIL FOR 1953

President

C. T. INGOLD, D.Sc., F.L.S.

Vice-Presidents

C. J. HICKMAN, Ph.D.
P. H. GREGORY, D.Sc., F.L.S.
A. A. PEARSON, F.L.S.

Secretary

J. WEBSTER, B.Sc.
Department of Botany, The University, Sheffield, 10

Foray Secretary

F. B. HORA, Ph.D.
Department of Botany, The University, Reading, Berks.

Treasurer

W. BUDDIN, M.A.
'Chiltern Court', St Peter's Avenue, Caversham, Reading, Berks.

Editors

P. H. GREGORY, D.Sc., F.L.S.
Rothamsted Experimental Station,
Harpenden, Herts.

G. C. AINSWORTH, Ph.D., F.L.S.
University College,
Exeter, Devon

Council

The Officers with the following in addition:

C. C. V. BATTS
P. W. BRIAN
C. L. DUDDINGTON
M. B. ELLIS
P. D. ORTON
J. T. PALMER
G. SMITH
I. F. STOREY
Mrs M. P. TOPPING

B. BARNES, Prof. W. BROWN, A. D. COTTON, Prof. Dame HELEN GWYNNE-VAUGHAN, J. RAMSBOTTOM, E. W. SWANTON, Miss E. M. WAKEFIELD, M. WILSON

PLANT PATHOLOGY COMMITTEE

Chairman

C. J. HICKMAN, Ph.D.

Secretary

C. C. V. BATTS, B.Sc., Ph.D.
National Institute of Agricultural Botany, Huntingdon Road, Cambridge

J. E. CROSSE
S. H. CROWDY
P. H. GREGORY
J. E. E. JENKINS
E. C. LARGE
E. LESTER
Miss M. NOBLE
N. F. ROBERTSON
Miss G. M. WATERHOUSE
P. H. WILLIAMS

The President and the Secretary *ex officio*

Recorder

(new British species and records)

M. B. ELLIS, B.Sc., Ph.D.
Commonwealth Mycological Institute, Ferry Lane, Kew, Surrey

The subscription price is 50*s.* per volume (post free), or $8.50 in U.S.A., payable in advance. Subscriptions may be sent to any Bookseller, or to the Cambridge University Press, Bentley House, 200 Euston Road, London, N.W. 1.

Enquiries from the U.S.A. should be addressed to the Cambridge University Press, American Branch, 32 East 57th Street, New York 22.

Vol. 36, Part 4 December 1953

THE NEW CODE OF BOTANICAL NOMENCLATURE

By G. C. AINSWORTH

Fungi, being traditionally regarded as plants, are subject to the regulations governing botanical nomenclature. The publication of the *International Code of Botanical Nomenclature** as adopted by the Seventh International Botanical Congress is, therefore, an event of importance not only to every contributor to these *Transactions* who wishes to use names 'correctly' or to propose new names, but also to the editors and to every mycologist, for mycology, like botany, 'cannot make satisfactory progress without a precise system of nomenclature which is used by the great majority of [mycologists] in all countries' (Art. 1). Professional systematists will read, and frequently re-read, the new Code with close attention, while the finer points of interpretation will test the judgement of experienced nomenclaturists. This review is not for such specialists. It is merely an attempt to indicate some of the many important changes in the Code which are of general interest to mycologists and to draw attention to certain requirements of the Code which are frequently overlooked.

General Considerations

The Code, as heretofore, is divided into principles (Arts. 1–9, 11–22), rules (22–83) and recommendations which are set out in a series of 83 Articles. The recommendations, previously numbered as a separate series, are now designated by the addition of letters to the number of the Article to which they relate.

According to Art. 4, 'the essential points of nomenclature are (1) to aim at fixity of names' and '(2) to avoid or to reject the use of forms and names which may cause error or ambiguity or throw science into confusion'. Botanical nomenclature thus deals with the *terms* which denote the categories of taxonomic groups or *taxa* (sing. *taxon*) and the *names* which are applied to taxa (Art. 8) and these objectives are achieved by the use of nomenclatural types and by the law of priority, but it should be noted that 'the Rules of priority and typification do not apply to names of taxa above the rank of order' (Art. 26).

A new Art. 10 distinguishes between *legitimate* and *illegitimate* names or epithets, names or epithets in accordance with or contrary to the Rules,

---

* *International Code of Botanical Nomenclature* adopted by the Seventh International Botanical Congress, Stockholm, July 1950. Prepared by J. Lanjouw (chief editor), Ch. Baehni, E. D. Merrill, H. W. Rickett, W. Robyns, T. A. Sprague (members of the editorial committee), F. A. Stafleu (secretary of the committee). Avec une traduction française par Ch. Baehni. Pp. 228. (Utrecht, Netherlands: International Bureau for Plant Taxonomy and Nomenclature of the International Association for Plant Taxonomy, September 1952.) Price Dfl. 14 (Buckram Dfl. 15.90). [Bound copies may be obtained for 30*s*., post free, from Mr A. A. Bullock, The Herbarium, Royal Botanic Gardens, Kew, Surrey.]

Vol. 36, *Part* 3: *issued* 26 *September* 1953.

respectively, from the *correct name*, the name which must be adopted under the Code for a taxon with 'a particular circumscription, position and rank'. This last ensures that a fungus can have as many correct names as there are different views on its taxonomy. The same article also distinguishes between *effective* publication (by the distribution of printed matter, but not after 1 Jan. 1953 as tradesmen's catalogues or newspapers, Art. 39) and *valid* publication which is effective publication conforming to the special conditions set out in Arts. 42–54. Of these, Art. 44 (the old Art. 38) states: 'On and from 1 Jan. 1953, names of new taxa of recent plants, the bacteria excepted, are considered as validly published only when they are accompanied by a Latin diagnosis.'

The sequence of the familiar categories from division to species (with such intermediate or subspecific categories as the situation requires) is unchanged, but the Code now clearly states that the relative order of the categories as specified in Arts. 12–14 must not be changed and that names denoting misplaced categories are not validly published. An exception, however, is made for the names of the subdivision of genera used by Fries in the *Systema mycologicum* which are to be treated as validly published, although Fries termed them 'tribes', a term which is correctly applied only to a subdivision of a family. The rules that the names of orders should be taken from that of the type family, with the ending *-ales* (Art. 27) and that of a family from the type genus, with the ending *-aceae* (Art. 28) are familiar but there is a new recommendation (Rec. 26*a*) that the names of divisions should end in *-phyta* except those of Fungi which should end in *-mycota* (i.e. Eumycota not Eumycetes, Myxomycota not Myxomycetes, etc.; *-mycetes* being the ending for the names of classes of Fungi; *-mycetidae*, that for subclasses).

Names of pleomorphic fungi

The one article specially inserted in the Code for the convenience of mycologists, Art. 69 (the old Art. 57), has been redrafted and merits quotation in full:

> In Ascomycetes and Basidiomycetes with two or more states in the life cycle (except those which are lichen-fungi), but not in Phycomycetes, the first valid name or epithet applied to the perfect state takes precedence. The perfect state is that which bears asci in the Ascomycetes, which consists of the spores giving rise to the basidia in the Uredinales and of the chlamydospores in the Ustilaginales, or which bears basidia in the remaining Basidiomycetes. The type specimen of a state must bear that state. However, the provisions of this article shall not be construed as preventing the use of names of imperfect states in works referring to such states. The author who first describes a perfect state may adopt the specific epithet of the corresponding imperfect state, but his binomial for the perfect state is to be attributed to him alone, and is not to be regarded as a transfer.
>
> When not already available, binomials for imperfect states may be proposed at the time of publication of a perfect state or later, and may contain either the specific epithet of the perfect state or any other epithet available.

Comment on this clear and unequivocal statement is hardly necessary. The new article follows very closely the wording suggested by G. R. Bisby (*Mycologia*, **36**, 283, 1946) and is in line with prevailing practice.

STARTING-POINTS

The application of the principle of priority is given greater precision by specifying the exact dates for the starting-points of the nomenclature for different groups, Lichenes and Myxomycota, 1 May 1753 (Linnaeus, *Species plantarum* ed. 1); Uredinales, Ustilaginales and Gasteromycetes, 31 Dec. 1801 (Persoon, *Synopsis methodica fungorum*); Fungi caeteri, 1 Jan. 1821 (Fries, *Systema mycologicum*, Vol. 1). Art. 22 goes on to state that the *Elenchus fungorum*, 1828, is to be treated as part of the *Systema* and that 'names of FUNGI CAETERI published in other works between the dates of the first (Vol. 1) and last (Vol. 3, part 2 and index) parts of the *Systema* which are synonyms or homonyms of names of any of the FUNGI CAETERI included in the *Systema* do not affect the nomenclatural status of names used by Fries in this work', a somewhat technical but not unimportant point.

NEW SPECIES

The application of names of taxa is determined by means of nomenclatural types, and in Art. 18 the various kinds of types are defined, while Art. 21 states that the nomenclatural type of a species or taxon below the rank of species is 'a single specimen or other element except...for most non-vascular plants the type may consist of more than one individual, which ought to be conserved permanently and assembled on one herbarium sheet or preparation'. This article also has notes to the effect that when it is impossible to preserve a type specimen and for species without a type specimen the type may be a figure and/or a description.

Among the miscellaneous recommendations which accompany Art. 54 the following points especially merit the attention of those who may wish to propose new species or subspecific taxa: the type specimen should be indicated and also the place where it is preserved (Rec. 54A); the description of a parasitic fungus should always be followed by an indication of the host which should be designated by the Latin scientific name (Rec. 54G); the description should be accompanied by figures with details of structure as an aid to identification; the explanation of the figures should indicate the specimen(s) on which they are based (Rec. 54F); and lastly, the etymology of new generic names and specific epithets should be given when the meaning may not be obvious (Rec. 54H). It may also be appropriately noted here that the new Code recommends that all specific and infra-specific epithets should now be written with a small initial letter, although botanists like zoologists apparently still feel unable to aline themselves completely with bacteriologists, for authors who so desire may still use capital initial letters in certain cases (Rec. 82G).

Finally, it may be noted that only the metric system should be used in botany for reckoning weights and measures (foot, inch,...pound, ounce... should be rigorously excluded from scientific language, Rec. 83D), temperatures should be expressed in degrees of the centigrade thermometer of Celsius (Rec. 83G), and authors should indicate clearly and precisely the scale of the figures which they publish (Rec. 83F).

VARIETIES

According to Art. 34: 'For nomenclatural purposes, a species or any taxon below the rank of a species is regarded as the sum of its lower taxa, if any' and further 'The description of a subordinated taxon which does not include the nomenclatural type of the higher taxon automatically creates a second subordinated taxon of the same rank which has as its nomenclatural type the type of the higher taxon.' This second taxon, the one that includes the nomenclatural type of the next higher taxon, is designated by repeating the epithet of the higher taxon unaltered but, contrary to Art. 55, without citation of an author's name (Art. 35). For example, the recent proposal in these *Transactions* of *Hydnangium carneum* Wallr. var. *xanthosporum* Hawker automatically created a variety *Hydnangium carneum* Wallr. var. *carneum* based on the type of Wallroth's species, and the use of the binomial *Hydnangium carneum* would now cover both varieties.

NEW COMBINATIONS

'On and after 1 Jan. 1953, new transfers or new combinations,...will be considered validly published only when the basonym (name-bringing or epithet-bringing synonym) is clearly indicated with its author and the place and date of publication' (Art. 42). Example: '*Ceratosporella stipitata* (Goid.) Hughes comb. nov. ≡ *Speira stipitata* Goidanich, *Malpighia*, **12**, 38, 1934' (*Trans. Brit. mycol. Soc.* **35**, 243, 1952). The editors would be grateful if contributors would bear this recommendation in mind and also remember in this connexion the instruction of Rec. 17A that 'changes in nomenclature should only be made after adequate taxonomic study'.

SYNONYMS

There are many pitfalls in the citation of rejected names, and one of the most frequent causes of misunderstanding is the citation, without comment, of an interpretation of what the author of a name intended. Names cited in synonymy should be spelt exactly as published by their author; any explanatory words required being inserted in brackets. If a name is adopted as valid with alterations from the form as originally published, it is desirable that in full citations the exact original form should be appended (Rec. 60H), e.g. *Ustilago striiformis* (Westend.) Niessl syn. *Tilletia striiformis* (Westend.) Saccardo, 1877 (as *T. striaeformis* (Westend.) Niessl).

Also, it should always be remembered that *misidentifications* should not be cited in the synonymy but added after it and a misapplied name should be indicated by the words '*auct. non*' followed by the name of the original author and the bibliographical references (Rec. 60F).

CONSERVATION

In order to prevent the undesirable consequences which sometimes follow the rigid application of the rules, and particularly of the principle of priority to the nomenclature of genera families, orders and intermediate taxa the Code provides lists of names which must be retained as exceptions (Art. 24). Such names are said to be 'conserved', a fact which should on formal occasions be indicated by the addition of 'nom. conserv.' after the author citation, e.g. *Marasmius* Fr. nom. conserv., which is conserved against the earlier name *Microphale* S. F. Gray. The final authority for the conservation of names is an International Botanical Congress, but when a name proposed for conservation has been approved by the Advisory Board and General Committee, it may then be retained pending the decision of Congress (Art. 25). International Congresses have always set their face against the conservation of specific names, but it seems not unlikely that before long a Congress may agree to the principle of the rejection of specific names which would achieve the same objective as conservation of specific names without many of the disadvantages (see *Taxon*, **1**, 78–80, 1952).

The first collected list of proposals for the conservation of generic names of fungi appeared in 1930, twenty-five years after the principle of *nomina generica conservanda* had been accepted by the Vienna Congress. No action was taken on any of these names at the 1935 Congress and then war intervened. Just before the outbreak of war a committee of the British Mycological Society set an example by examining the evidence for and against conservation of a number of these names and published its findings (*Trans. Brit. mycol. Soc.* **23**, 215–232, 281–292, 1939). Partly as a result of this work the Stockholm Congress agreed to the conservation of thirteen generic names of fungi and these are published in Appendix V of the new Code (pp. 76–78).

Names proposed for conservation are examined before submission to the Advisory Board and General Committee by the Special Committee for Fungi (Secretary: Donald P. Rogers, New York Botanical Garden, Bronx Park, New York 58, N.Y., U.S.A.), and this committee has recently distributed details of its proposals to the Advisory Board regarding a number of additional generic names of fungi (see *Taxon*, **2**, 29–32, 1953; *Mycologia*, **45**, 312–322, 1953 for the full text). A further sixteen generic names (including *Candida* Berkh., *Phoma* Sacc., and *Urocystis* Rabenh.) are recommended for conservation while for others conservation is for various reasons not recommended.

ANNOTATED LIST OF BRITISH AQUATIC CHYTRIDS

By HILDA M. CANTER*

Freshwater Biological Association, Ferry House, Ambleside, Westmorland

Notes and records are given of species of aquatic chytrids occurring in the British Isles, listed under the classification of Sparrow (1943). Information on the whereabouts of reference material is given and a host and substratum index is appended.

INTRODUCTION

The first list of British Chytrids, compiled by Massee (1891), contained six species: *Rhizidium westii*, *R. intestinum*, *Reesia amoeboides*, *Chytridium helioformis*, *Olpidium lemnae* and *Polyphagus euglenae*. A second list with notes on unsatisfactory records and on nomenclature was published by Ramsbottom (1916*a*, *b*). The eighteen species listed were: *Olpidium sphacellarum*, *O. tumefaciens*, *O. plumulae*, *O. endogenum*, *O. lemnae*, *O. gillii*, *Ectrogella bacillariacearum*, *Pleolpidium cuculus*, *P. irregulare*, *Woronina polycystis*, *Rhizophidium barkerianum*, *R. eudorinae*, *Phlyctochytrium westii*, *Entophlyctis helioformis*, *Diplophlyctis intestina*, *Polyphagus euglenae*, *Resticularia boodlei* and *R. nodosa*. Only seven of these still remain as records in the Chytridiales, and two of these are placed in a different genus. Two others remain as imperfectly known species. The remainder, except for *Resticularia nodosa* and *R. boodlei*, find places in the Saprolegniales, Lagenidiales, Plasmodiophorales and Hyphochytriaceae. *R. nodosa* Dang. and *R. boodlei* Fritsch, as described by Fritsch (1903), are discussed in detail by Ingold (1949) who says: 'It is clear that *R. boodlei* shows no feature which suggests that it belongs to the Phycomycetes and the same is true of the fungus which Fritsch identified as *R. nodosa*.' Although seven terrestrial members of the Synchytriaceae are recorded by Ramsbottom, no aquatic species was then known. However, it now seems possible that the bodies occurring in members of the Conjugales which were described by the algologists Thwaites (1846–7), Shadbolt (1852), and Smith (1853), are prosori of the aquatic genera *Micromyces* and *Micromycopsis*. Species of *Physoderma* and *Urophlyctis* recorded by Ramsbottom (1916*a*) are not considered here.

After 1916 the aquatic chytrids received little attention, until the survey made by Sparrow (1936) was followed by a series of papers by Ingold (1940, 1941, 1944), Canter (1946, 1947*a*, *b*, 1949*a–d*, 1950*a–d*, 1951) and Canter & Lund (1948, 1951), all of which were mainly studies of species living on algae. The study of chytrids utilizing chitin, keratin or cellulose, and those parasitizing other aquatic fungi remains an open field. Knowledge of the Chytridiales is growing rapidly, and ideas on classification are changing. Many more studies must be made before a satisfactory system of classification can be evolved. The most recent is that of Whiffen (1944). However, it is simpler, especially for the student, to follow that given in Sparrow's (1943) comprehensive book.

---

* Mrs J. W. G. Lund.

The Chytridiales in the early classification included simple aquatic Phycomycetes possessing one or two flagella. Following Sparrow (1943) the Chytridiales as here understood is restricted to 'Chytrids' with posteriorly uniflagellate zoospores formed in a sporangium. Biflagellate species such as *Ectrogella bacillariacearum* Zopf, *Lagenidium gracile* Zopf, *L. rabenhorstii* Zopf, *Pseudolpidium fusiforme* Fischer and *Woronina polycystis* Cornu, which have been recorded for this country as Chytridiales, are not discussed.

The list is divided into families and genera with the species arranged alphabetically within each genus. Doubtful species and records are included among the good ones. The excluded species have been grouped together and are listed on pp. 294-5. Synonyms are given only where the fungus has been recorded for Great Britain under that name. Relevant notes and citations from other writers appear where necessary. In the reference to the paper where a fungus is described an asterisk (*) denotes a figure. This is not indicated for excluded species. New records made by the writer are indicated as 'Canter (unpublished)'. Where possible the most precise location is given. Further details of the locality may appear on p. 296, where an index to the site numbers used by Sparrow (1936) can also be found. Sometimes, e.g. records by Cooke (1882-4) in *British Freshwater Algae* and Massee (1891) in *British Fungi, Phycomycetes and Ustilagineae*, the locality is unknown. In describing the habit of the fungus the word 'parasitic' has been omitted, so 'on'...or 'in'...can be taken to mean parasitic unless otherwise stated. This list covers all species published up to the end of December 1952.

There is no type collection of chytrids in this country. Good preservation of these organisms is a difficult problem. It seems that each species has to be treated individually, and the reactions of an alga to the preservative have to be considered as well. Preserved material of many of the species I have described is lodged at the Freshwater Biological Association's Windermere Laboratory, Westmorland, England, and communication with F. K. Sparrow reveals that he has a large collection of slides from Great Britain.

My best thanks are due to Drs J. W. G. Lund, P. H. Gregory and G. C. Ainsworth and Prof. C. T. Ingold for all the helpful advice they have given during the preparation of this work.

OLPIDIACEAE (Aquatic)

Olpidium endogenum (Braun) Schroeter

Archer (1860*): in *Closterium lunula* Ehrenb., no locality given. The description of this fungus is very meagre and it may possibly be a species of *Myzocytium*, in spite of the fact that the individual plants are stated by Archer to be separate. Neither the flagellation of the swarmers nor resting spores were observed.

Cooke (1882–4*): in *Closterium lunula*, *Vaucheria*, etc., no locality given. The flagellation of the zoospores is not stated and resting spores were not observed. Cooke (1882–4, p. 198) suggests that this is the species figured by Henfrey (1859*) in *Eremosphaera viridis* (*Chlorosphaera oliveri*) from Prestwick Carr, Northumberland. This seems unlikely as the discharge tubes of the latter possess no endobiotic swelling.

Ramsbottom (1916*a*).

Sparrow (1936*): in *Mougeotia* sp. and *Zygnema* sp., site 1, Cambridge. Sparrow (1936, p. 427) doubts whether or not these two fungi can be assigned to the species *Olpidium endogenum*. He states that they closely resemble *O. spirogyrae* Skvortzow (1925) and the unnamed species in *Spirogyra* illustrated by Gwynne-Vaughan & Barnes (1937*). A somewhat similar parasite was found by Sparrow (1936*) in *Spirogyra*, site 3, Cambridge, but no zoospores were observed.

Olpidium entophytum (Braun) Rabenhorst

Sparrow (1936*): in *Cladophora* sp., Hobson's Conduit, Trumpington St., Cambridge. The fungus described by Henfrey (1859*) in *Eremosphaera viridis* de Bary from Prestwick Carr, Northumberland, may belong here, but the description is far too inadequate to draw any definite conclusions.

Olpidium euglenae Dangeard

Sparrow (1936*): in *Euglena* sp., site 4, Cambridge. Sparrow (1936, p. 428), states the need for further data on the method of zoospore discharge and zoospores before a final identification is possible.

Olpidium gregarium (Nowak.) Schroeter

Sparrow (1936*): in rotifer eggs, site 4, Cambridge.

Olpidium hyalothecae Scherffel

Canter (1949*b**): in *Hyalotheca dissiliens* (Sm.) Bréb., Blelham Tarn Bog near Wray Castle, and Lake shore, Esthwaite Water, near Hawkshead, Lancs.

(?) **Olpidium plumulae** (Cohn) Fischer (≡ *Chytridium plumulae* Cohn)

Magnus (1872 as *C. plumulae*): in preparations of antheridial specimens of *Callithamnion plumula* Cohn [*Antithamnion plumula* (Ellis) Thur.], Plymouth, Devonshire (coll. Kny). Murray (1893, and extract of this paper by Holmes (1893), as *C. plumulae*): in *Callithamnion* sp., coasts of Britain. Ramsbottom (1916*a*), Smith & Ramsbottom (1916).

Sparrow (1943, p. 99) says: 'The identity of this fungus cannot be determined without further observations on the zoospores. It may be *Eurychasmidium tumefaciens* (Magnus) Sparrow.'

Olpidium utriculiforme Scherffel

Canter (1949*b**): in *Closterium lunula* Ehrenb., *C. costatum* Corda and *C. dianeae* Ehrenb., from Blelham Tarn Bog, near Wray Castle, Lancs.

Rozella cuculus (Butler) Sparrow (≡*Pleolpidium cuculus* Butler)

Butler (1907*, as *P. cuculus*): in sporangia of *Pythium intermedium* de Bary, Cork, Ireland. Ramsbottom (1916*a*) and Smith & Ramsbottom (1916) as *P. cuculus.*

Rozella irregularis (Butler) Sparrow (≡*Pleolpidium irregulare* Butler)

Butler (1907* as *P. irregulare*): in *Pythium* (?) *vexans*, Kew, Surrey. Ramsbottom (1916*a*) and Smith & Ramsbottom (1916) as *P. irregulare.*

Rozella polyphagi (Sparrow) Sparrow (≡*Pleolpidium polyphagi* Sparrow (1933))

Sparrow (1933, p. 21, as *Pleolpidium polyphagi*; 1936* as *P.* (*Rozella*) *polyphagi*): in prosporangia of *Polyphagus euglenae* Nowak., ditch, rear of Leys School, Cambridge (type locality).

The fungus described as *P. euglenae* from this locality was designated by Sparrow (1943, p. 300) as *P. euglenae* var. *minor* Nowak., now *P. laevis* (Nowak.) Bartsch. Sparrow found a few specimens with aculeolate zygospores in this locality which, according to Bartsch (1945), would belong to *P. euglenae* Nowak. emend. Bartsch. As the host in this record was collected on the same day and at the same place as *P. laevis* and *P. euglenae* Nowak. emend. Bartsch it may in fact be either or both these species.

? (Rozella) septigena Cornu

Sparrow (1936): in *Saprolegnia* spp. and *Achlya* spp., Barton Mills, Suffolk. Zoospores were not observed and therefore the identification is subject to doubt (see Sparrow 1943, p. 121).

Rozella sp.

Canter (1950*d**): in sporangia of *Chytridium oedogonii* Couch, Blelham Tarn.

(?) Rozella sp.

Sparrow (1936, as (?) *Pseudolpidium pythii* Butler): in *Pythium* sp. isolated from heathland sand, Mildenhall, Suffolk. Sparrow (1936, p. 425) states that the fungus is identical with that figured by Butler (now *Olpidiopsis pythii* (Butler) Karling), but since no zoospores were observed it may be a species of *Rozella.*

(?) Pleotrachelus petersenii Lund

Sparrow (1936*): in *Zygnema* sp., site 1, Cambridge. Sparrow (1936, p. 428) suggests that since no zoospores were observed this fungus may be a species of *Petersenia* or *Lagenidium.*

ACHLYOGETONACEAE

Septolpidium lineare Sparrow

Sparrow (1933, p. 215; 1936*): in *Synedra* sp., site 1 (type locality) and site 3, Cambridge, and site 5, Surrey.

SYNCHYTRIACEAE (Aquatic)

Endodesmidium formosum Canter

Canter (1949*c**): in *Netrium oblongum* (de Bary) Lütkem, *Cylindrocystis crassa* de Bary and *C. brebissonii* Menegh., Rusland Moss.

Micromycopsis fischeri Scherffel

Canter (1949*c**): in *Tetmemorus brebissonii* (Menegh.) Ralfs, from a path leading to Three Dubs Tarn, Claife Heights, Near Sawrey, Lancs.

Micromycopsis intermedia Canter

Canter (1949*c**): in *Zygnema* spp., Blelham Tarn Bog, near Wray Castle.

Micromycopsis mirabilis Canter

Canter (1949*c**): in *Closterium lunula* Ehrenb., *C. dianae* Ehrenb., *C. costatum* Corda, *C. kutzingii* Bréb., and *Closterium* sp., from Blelham Tarn Bog, near Wray Castle.

Micromyces laevis Canter

Canter (1949*c**): in *Mougeotia* sp., Clay Pond, near Wray Castle.

Micromyces petersenii Scherffel

Canter (1949*c**): in *Mougeotia* sp., Blelham Tarn Bog, near Wray Castle.

Micromyces zygogonii Dangeard

Canter (1949*c**): in *Spirogyra* sp., Clay Pond, near Wray Castle.

A form of *M. zygogonii* was found in *Mougeotia* sp. in Blelham Tarn Bog, near Wray Castle (Canter, 1949*c*, p. 84).

'ASTEROSPHERES'

Thwaites (1846–7), Shadbolt (1852) and Smith (1853) refer to spiny bodies (asterospheres), causing hypertrophy in members of the Conjugales, which may well have been prosori of *Micromyces* or *Micromycopsis* spp.

PHLYCTIDIACEAE

Phlyctidium apophysatum Canter

Canter (1947*a**): on *Mougeotia* sp., Clay Pond, near Wray Castle. Further investigations on material from the tarn at Tarn near Silloth, Cumberland, on *Mougeotia* sp. suggest that this fungus exhibits endo-exogenous development.

Phlyctidium chlorogonii Serbinow

Scourfield (1936*): on *Chlorogonium elongatum* Dang., Epping Forest, Essex. Whether this identification is correct must await further investigations as the rhizoid described by Serbinow is an inconspicuous knob-like structure, whereas in the one instance Scourfield could see the rhizoid it was threadlike, resembling species of the genus *Rhizophidium*.

Rhizophidium acuforme (Zopf) Fischer

Grove (1917*): on *Chlamydomonas intermedia* Chodat, Harborne, Warwickshire. The sporangia of this fungus resemble those figured by Zopf, but owing to slight differences in the rhizoidal system and to the lack of resting spores this identification remains uncertain.

Rhizophidium ampullaceum (Braun) Fischer (≡ *Olpidium ampullaceum* (Braun) Rabenhorst)

Cooke (1882–4* as *Olpidium ampullaceum*): on various algae, the host in Pl. LXXXI, fig. 3, is given as *Mougeotia* sp., locality unknown. W. R. I. Cook (1932*): on *Oedogonium crassusculum* Wittr. var. *idiosporum* Nordst et Wittr., from Rhythfyn, near Aberystwyth, Wales. W. R. I. Cook (1936): Glamorgan, Wales. Cooke (1882–4, p. 198) suggests that this is the fungus figured by Henfrey (1859, figs. 13, 14) on *Eremosphaera viridis* de Bary; Prestwick Carr, Northumberland.

Rhizophidium anomalum Canter

Canter (1950*a**): on *Apiocystis brauniana* Naeg., lake shore path, High Wray Bay, Windermere.

Rhizophidium barkerianum (Archer) Rabenhorst (≡ *Chytridium barkerianum* Archer)

Archer (1867, as *C. barkerianum*): on *Zygnema* sp., Callery Bog, Ireland. Ramsbottom (1916*a*); Smith & Ramsbottom (1916). Sparrow (1943, p. 216) considers this fungus to be of doubtful validity.

Rhizophidium carpophilum (Zopf) Fischer

Sparrow (1936*): on eggs of *Achlya* spp. and *Monoblepharis macrandra* (Lagerheim) Woronin, from site 2, Cambridge; and on eggs of *Dictyuchus monosporus* Leitgeb., from Kew Gardens, Surrey.

Buckley & Clapham (1929*) have assigned with reserve to *Rhizophidium carpophilum* (Zopf) Fischer a fungus on eggs of *Dibothriocephalus latus* (a helminth) from the water tank supplying the laboratory tap water, Department of Helminthology, School of Hygiene and Tropical Medicine, London, which according to Sparrow (1943, p. 189) may be referable to *Rhizophidium vermicola*.

Rhizophidium columnaris Canter

Canter (1947*b**): on *Spirotaenia condensata* Bréb., Clay Pond, near Wray Castle.

Rhizophidium ephippium Canter

Canter (1950*c**): on *Stylosphaeridium stipitatum* (Bachm.) Geitler, Windermere (north and south basins) and Esthwaite Water. Canter (unpublished): Loweswater.

Rhizophidium eudorinae Hood

Hood (1910*): on *Eudorina elegans* Ehrenb.. Bracebridge Pool, Sutton Park, Warwickshire. Ramsbottom (1916*a*); Smith & Ramsbottom (1916).

It is suggested by Canter (1946, p. 134) that *R. eudorinae* represents material of *Dangeardia mammillata* together with an epibiotic chytrid belonging possibly to *Rhizophidium* or *Chytridium*.

Rhizophidium fragilariae Canter

Canter (1950*c**): on *Fragilaria crotonensis* (A. M. Edw.) Kitton, in Windermere (north and south basins) and Esthwaite Water. Canter (unpublished): in Bassenthwaite Lake and Ullswater.

Rhizophidium fulgens Canter

Canter (1951*): on *Gemellicystis neglecta* (Teiling) Skuja in Loweswater and Crummock Water, and in Swithland Reservoir, Leicestershire. Canter (unpublished): in Bassenthwaite Lake and Hatch Mere (coll. E. M. Lind).

Rhizophidium fusus (Zopf) Fischer

Sparrow (1943, p. 202): on *Melosira varians* Ag., from site 1, and Coe Fen, Cambridge, and site 5, Surrey.

This fungus was originally identified by Sparrow (1936*) as *Rhizophidium lagenula* (Br.) Fischer. Under this species Sparrow (1936*) also describes a form with more broadly fusiform sporangia often resting on a broad base, on *Synedra* sp. and *Melosira* sp. from Coe Fen, Cambridge.

Rhizophidium globosum (Braun) Rabenhorst

W. R. I. Cook (1932*): on *Ulothrix* sp., Graves Park, Sheffield. W. R. I. Cook (1936): Glamorgan, Wales. A doubtful record is given by Sparrow (1936*) on a rotifer, site 2, Cambridge.

Rhizophidium goniosporum Scherffel

Sparrow (1936*): on *Tribonema bombycina* (Ag.) Derb. et Sol. forma *minor*, from sites 1 and 4, Cambridge.

Rhizophidium hyalobryonis Canter

Canter (1951*): on *Hyalobryon mucicola* (?) Pascher (now *H. polymorphum* Lund.) Loweswater. Canter (unpublished): Crummock Water, Blelham Tarn and Loughrigg Tarn.

Rhizophidium laterale (Braun) Rabenhorst

Ingold in communication to Sparrow (see Sparrow 1943, p. 170): on a thin-walled *Ulothrix*, England. Communication with Prof. Ingold gives the locality as 'near Leicester'. This identification remains doubtful as the form above lacked both rhizoids and hemispherical cyst.

Rhizophidium megarrhizum Sparrow

Sparrow (1943, p. 171). This fungus was originally identified by Sparrow (1936*) as *Rhizophidium subangulosum*; on *Oscillatoria* sp., in a ditch at Leys

School, Cambridge. Canter & Lund (1951*): on *Oscillatoria agardhii* Gom. var. *isothrix* Skuja, Windermere (south basin), Esthwaite Water and Loughrigg Tarn. Ingold in Canter & Lund (1951* p. 368): on *Oscillatoria* sp. in Bradbourne Park Lake, Riverhead, Sevenoaks, Kent. Canter (unpublished): on *Oscillatoria* sp., Bigland Tarn, near Greenodd, Lancs.

Rhizophidium planktonicum Canter

Canter & Lund (1948*): on *Asterionella formosa* Hass., Windermere (north and south basins), Esthwaite Water and Blelham Tarn. Canter (unpublished): Loweswater, Crummock Water, Ullswater, Bassenthwaite Lake, Hawes Water and Loughrigg Tarn. Further studies indicate that this is possibly an aggregate species (Canter & Lund, 1951, p. 361). The fungus found by Prof. C. T. Ingold on *Asterionella* sp., from Kilby Bridge, Leicestershire, and mentioned in (Canter & Lund, 1948, p. 259) cannot definitely be assigned to *Rhizophidium planktonicum* as resting spores were not observed.

Rhizophidium simplex (Dang.) Fischer

Sparrow (1936*): on *Spirogyra* sp., from site 1, Cambridge, and on *Chlorococcum* (?) from site 5, Surrey.

Rhizophidium sphaerocystidis Canter

Canter (1950*c**): on *Sphaerocystis schroeteri* Chodat, Windermere (north and south basins), Blelham Tarn and Esthwaite Water. Canter (unpublished): Buttermere and Crummock Water; on *Dictyosphaerium pulchellum* Wood, Windermere (north and south basins).

Rhizophidium sphaerotheca Zopf

Sparrow (1943, p. 173). Sparrow (1936*, as a form of *Rhizophidium globosum*): on *Typha* pollen, site 2, Cambridge.

Rhizophidium subangulosum (Braun) Rabenhorst

Sparrow (1943, p. 170). Sparrow (1936*, as a form of *Rhizophidium globosum*): on *Oscillatoria* sp., from site 1, Cambridge.

Rhizophidium transversum (Braun) Rabenhorst

W. R. I. Cook (1932*): on *Eudorina elegans* Ehrenb., Epping Forest, Essex. Sparrow (1943, p. 199) comments that the sporangia are too spherical for *R. transversum* and that the fungus may be *R. acuforme* (Zopf) Fischer.

Rhizophidium vermicola Sparrow

Sparrow (1943, p. 189). Sparrow (1936*, as *Rhizophidium sphaerocarpum* (Zopf) Fischer): parasitic (?) on a nematode infected by other fungi, from site 4, Cambridge.

It is suggested by Sparrow (1943, p. 189) that the fungus described as *Rhizophidium carpophilum* by Buckley & Clapham (1929*) on helminth eggs may be referable to *R. vermicola*.

Rhizophidium zoophthorum (Dang.) Fischer

Sparrow (1943, p. 195): coll. J. Bayley Butler, on liver fluke eggs, Ireland.

Rhizophidium sp.

Canter (1947*a**): on sporelings of *Oedogonium* sp., in Clissold Park Lake, London.

? Rhizophidium or **Chytridium** spp.

The following are records of fungi which remain unclassified as the method of zoospore discharge was not seen.

Sparrow (1936, p. 438): on the internodal cells of *Nitella tenuissima* (?), Wicken Fen, Cambridge. Sparrow states that in general the fungus resembles *Phlyctidium chlorogonii* Serb. Sparrow (1936*, p. 438): on a spherical green alga, from site 4, Cambridge. Sparrow found both large pyriform sporangia with a rhizoidal system which appeared to be knob-like and similar but smaller sporangia with a branched rhizoidal system in the same collection.

Sparrow (1936,* p. 443): on *Spirogyra* sp., Coe Fen, Cambridge. See also Sparrow (1943, p. 213).

Sparrow (1936,* p. 443): on *Spirogyra* sp., from site 1, Cambridge.

Grenfell (1894): two fungi on *Stauroneis phoenicenteron* (Nitzsch) Ehrenb. and (?) *Navicula sphaerophora* Kütz, in the Botanical Gardens, Regents Park, London. Grenfell states that the general appearance of the two fungi occurring on *S. phoenicenteron* and (?) *N. sphaerophora* is similar to *Septocarpus corynephorus* Zopf (*Podochytrium clavatum* Pfitzer), figured by Karop (1889), but that the lower portion of the sporangium is not cut off by a septum.

Reynolds (1940*): on *Staurastrum paradoxum* Meyen; Swithland Reservoir, Leicestershire.

Phlyctochytrium (?) biporosum Couch

Sparrow (1936*): on *Spirogyra* sp., from site 1, Cambridge. Sparrow (1943, p. 228), suggests that the fungus resembles in some respects *Rhizophidium haynaldii* (Schaarschmidt) Fischer and *R. rostellatum* (de Wildeman) Fischer.

Phlyctochytrium laterale Sparrow

Sparrow (1936*): on *Spirogyra* sp., from site 1, Cambridge.

Phlyctochytrium mucronatum Canter

Canter (1949*d**): saprophytic on *Closterium pritchardianum* Arch., the Clay Pond; and on *C. costatum* Corda, Blelham Tarn Bog, both localities near Wray Castle.

Phlyctochytrium proliferum Ingold

Ingold (1941*): on *Chlamydomonas* sp., in a temporary pond, Mortimer Common, near Reading, Berks.

Phlyctochytrium quadricorne (de Bary) Schroeter

Sparrow (1936*): on *Cladophora* sp., Trumpington Street Brook, Cambridge.

Rhizidiopsis emmanuelensis Sparrow

Sparrow (1933, p. 216; 1936*): on *Melosira varians* Ag. and *Nitzschia* (?) sp., from site 1, Cambridge (type locality).

Podochytrium clavatum Pfitzer

Sparrow (1936*): on *Fragilaria* sp., from site 5, Surrey.

Podochytrium lanceolatum Sparrow

Sparrow (1933, p. 216; 1936*): on *Melosira varians* Ag., from site 1, Cambridge (type locality).

Scherffeliomyces appendiculatus (Zopf) Sparrow (≡*Rhizidium appendiculatum* Zopf).

W. R. I. Cook (1932,* as *R. appendiculatum*): on *Chlamydomonas* sp., Epping Forest, Essex. Sparrow (1943, p. 249) states that if the sequence of development of this fungus as described by Cook is correct, then it can neither be referred to *Scherffeliomyces* nor *Rhizidium.*

Scherffeliomyces parasitans (Sparrow) Sparrow (≡*Scherffelia parasitans* Sparrow)

Sparrow (1933, p. 127, as *Scherffelia parasitans*; 1936*): on resting cells of *Euglena* sp., from site 4, Cambridge (type locality).

Rhizosiphon anabaenae (Rodhe et Skuja) Canter

Canter (unpublished): on *Anabaena affinis* Lemm. var. *intermedia* Griffiths, and *A. spiroides* Kleb., Lough Oughter (coll. H. C. Gilson).

Rhizosiphon crassum Scherffel

Canter (1951*): on *Anabaena* sp., Loweswater; and on *A. spiroides* Kleb. var. *crassa* Lemm., Swithland Reservoir. Canter (unpublished): on *Anabaena circinalis* (?) (Kütz.) Hansg., Esthwaite Water, Windermere (south basin), and Lough Oughter: on *A. affinis* Lemm. var. *intermedia* Griffiths, Loch Oughter (coll. H. C. Gilson) and Llyn Coron (coll. B. M. Griffiths): on *A. spiroides* Kleb., Loughs Oughter and Erne (coll. H. C. Gilson): on *Anabaena* sp., Hatch Mere (coll. E. D. Le Cren).

The fungus noted by Griffiths (1925); on *A. affinis* var. *intermedia* in Norton Mere, Albrighton, is identified by me as *Rhizosiphon crassum.*

Dangeardia mammillata Schröder

Ingold (1940): on *Eudorina elegans* Ehrenb., Swithland Reservoir. Canter (1946*): on *E. elegans*, Barn Elms Reservoir 5, London; Canter (1951): London Metropolitan Water Board Reservoirs, King George (Lea), and Walthamstow Race Course.

The fungus noted by Griffiths (1925) on *E. elegans* in Newton Mere is identified by Canter (1951, p. 153) as *Dangeardia mammillata*. Canter (1946, p. 133) suggests that *Rhizophidium eudorinae* Hood represents material of *D. mammillata* together with another epibiotic chytrid.

Septosperma anomala (Couch) Whiffen

Canter (1949*a**): on *Chytriomyces tabellariae* (Schroeter) Canter, Blelham Tarn Bog, near Wray Castle. Canter (1950*c**): on *Rhizophidium sphaerocystidis* Canter, Windermere (north and south basins). Canter (unpublished): on *Zygorhizidium willei* Löwenthal, Fisherty How, Low Wray Bay, Windermere; on sori of *Micromycopsis* sp., in a pool at 2000 ft., Grey Friar Mountain in the Coniston Range, Lancs.; on sporangia of *Rhizophidium planktonicum* agg., Sunbiggin Tarn.

'HYPERPARASITES'

Figures of incompletely known hyperparasites from the English Lake District are shown in Canter (1949*d*, fig. 2, *n–p*; 1950*d*, fig. 6, *z*).

Entophlyctis apiculata (Braun) Fischer

Sparrow (1936*): on resting cells of *Euglena* sp., from site 4, Cambridge. Sparrow (1936, p. 451) points out that since no rhizoids were observed the fungus might have been a species of *Olpidium*.

Entophlyctis aurea Haskins

Haskins (1946*): saprophytic in various grasses, wheat, oat and maize leaves, regenerated cellulose film, filter-paper and lens-paper; Cambridge.

Entophlyctis bulligera (Zopf) Fischer

Sparrow (1936*, described under *E. cienkowskiana* (Zopf) Fischer): immature stages of a fungus closely resembling *E. bulligera* were observed on *Bulbochaete* sp., Wicken Fen, Cambs.

†**Entophlyctis confervae-glomeratae** (Cienkowski) Sparrow

Sparrow (1943, p. 258). Originally described by Sparrow (1936*) under *E. cienkowskiana*: on *Spirogyra* sp., Wicken Fen and Streighton, Cambs; *Oedogonium* sp., from site 5, Surrey: *Cladophora* sp., Trumpington Street, Cambridge.

Entophlyctis helioformis (Dang.) Ramsbottom (≡ *Chytridium helioformis* Dang.)

Massee (1891, as *C. helioformis*): habit unknown, in the interior of *Chara*, *Nitella* and *Vaucheria*, locality not given. Ramsbottom (1916*a*, *b*).

† There appears to be some confusion regarding the algal hosts of these fungi. In the text in Sparrow (1936, p. 452), *E. rhizina* (?) is described from *Cladophora*, whereas both the legend to his figure (pl. 14, fig. 18), and the reference in Sparrow's book (1943, p. 259) give the alga as *Vaucheria*. This host was therefore presumed to be the correct one and communication with Dr F. K. Sparrow confirmed this belief. Thus in Sparrow (1936, p. 452), in line 17, *Cladophora* should be substituted for *Vaucheria* and in line 19, *Vaucheria* for *Cladophora*.

†**Entophlyctis rhizina** (Schenk) Minden

Sparrow (1936,* described under *E. cienkowskiana*): in *Vaucheria* sp., Streighton, Cambs. Sparrow (1943, p. 256) states that the fungus closely resembles that described by Schenk but the zoospore possesses a colourless instead of a coloured globule.

Entophlyctis sp.

Sparrow (1936,* described under *E. cienkowskiana*) on *Spirogyra* sp., from site 5, Surrey. Sparrow was unable to induce discharge of the zoospores and it remains a question whether the fungus is a species of *Entophlyctis* or *Endochytrium*.

Diplophlyctis intestina (Schenk) Schroeter (≡*Rhizidium intestinum* (Schenk) pro parte)

Massee (1891; as *R. intestinum*): habit unknown, on species of *Chara* and *Nitella*, locality not given. Ramsbottom (1916*a*, *b*). Richards (1951): in oat and maize leaves and stems used as bait, in the Water Garden, Roath Park, Cardiff, Wales.

I have found this fungus widely distributed, saprophytic, in *Nitella* sp., in the English Lake District.

Diplophlyctis laevis Sparrow

Richards (1951*): in moribund internodes of *Chara*; from a ditch in a railway cutting about a quarter of a mile east of Bridgend; and in fragments of oat leaf added to debris brought from Pysgodlyn Mawr, St Donat's, and from Kenfig Pool; all in Glamorgan, South Wales.

RHIZIDIACEAE

Rhizidium mycophilum Braun

Sparrow (1936, 1937): saprophytic on exuviae of Chironomidae, River Cam, Cambridge.

Rhizidium variabile Canter

Canter (1947*a**): saprophytic on *Spirogyra* sp., Chelsea Physic Garden pond, London.

Rhizidium windermerense Canter

Canter (1950*c**): on *Gemellicystis neglecta* (Teiling) Skuja; Blelham Tarn, Derwentwater, Esthwaite Water and Windermere (north and south basins). Canter (1951): Bassenthwaite Lake. Canter (unpublished): Loch Mhor in the Loch Ness basin (coll. C. H. Mortimer).

(?) **Rhizidium** spp.

Sparrow (1936*) describes two fungi saprophytic on decaying vegetable debris, from site 5, Surrey, which he considers may possibly be related to *Rhizidium*.

---

† See note on p. 288.

Rhizoclosmatium globosum H. E. Petersen emend. Sparrow

Sparrow (1936, 1937): saprophytic in midge integuments, River Cam, Cambridge.

I have found this fungus widely distributed, saprophytic, in insect exuviae in the English Lake District.

Asterophlyctis sarcoptoides H. E. Petersen

I have found this species widely distributed, saprophytic, in insect exuviae in the English Lake District.

Siphonaria variabilis H. E. Petersen

I have found this species widely distributed, saprophytic, in insect exuviae in the English Lake District.

Polyphagus euglenae Nowakowski emend. Bartsch (1945). (=*P. euglenae* Nowak. pro parte)

Massee (1891*): on *Euglena viridis* Ehrenb., locality not given. Wager (1913*): on *E. viridis*, at a sewage farm, Keighley, Yorks, Ramsbottom (1916*a*).

It seems likely that the few specimens with ornamented zygospores described by Sparrow (1936, p 453) on temporary resting cells of *Euglena* sp. in a ditch to the rear of Leys School, Cambridge, may belong here.

Other records of *Polyphagus euglenae* are listed below but in none of these is any description of the fungus given, therefore whether the record refers to *P. euglenae* Nowak. emend. Bartsch or *P. laevis* (Nowak.) Bartsch or both remains unknown.

Massee & Crossland (1905): on *Euglena viridis* Ehrenb., Seamer Moor, Scarborough in the north-east division of Yorks.

Mason (1927): on *Euglena* spp., Meanwood and neighbourhood, Leeds, mid-west division of Yorks.

Mason & Grainger (1947): vice-counties 62, 64 Yorks.

Polyphagus laevis (Nowakowski) Bartsch (=*P. euglenae* Nowakowski pro parte, *P. euglenae* var. *minor* Nowak.)

Wager (1913,* as *P. euglenae*): on *Euglena* sp., at a sewage farm, Keighley, Yorks. Sparrow (1936,* as *P. euglenae*; 1943, p. 300 as *P. euglenae* var. *minor*): on temporary resting cells of *Euglena* sp., in a ditch to the rear of Leys School, Cambridge.

It is possible that this species was present in Massee's material (1891) as he refers to smooth as well as minutely aculeolate resting spores.

Endocoenobium eudorinae Ingold

Ingold (1940*): in coenobia of *Eudorina elegans* Ehrenb., Swithland Reservoir, Leicestershire.

CLADOCHYTRIACEAE (Aquatic)

Cladochytrium hyalinum Berden

Haskins (1946*): saprophytic in grass, Cambridge.

Cladochytrium replicatum Karling

Sparrow (1936): saprophytic on leaves of *Elodea*, from site 2, and Wicken Fen, Cambs; on decayed grass leaves in a ditch, rear of Queens' College, Cambridge and site 5 Surrey; on decayed grass leaves and stems, Kew Gardens, Surrey.

CHYTRIDIACEAE

Chytridium acuminatum Braun

Cooke (1882–4*): on oogonia of *Oedogonium rothii* (Le Clerc) Pringsheim, locality not given.

Chytridium appressum Sparrow

Sparrow (1936*): on *Melosira varians* Ag., site 1, Cambridge.

Chytridium chaetophilum Scherffel

Sparrow (1936*): on *Bulbochaete* sp., Wicken Fen, Cambs.: saprophytic on *Typha* pollen (bait), site 5, Surrey: substratum unknown, coll. Odam, communication to Sparrow (see Sparrow, 1943, p. 339). Further details of the last record have been sent to the writer by Dr Odam. The fungus occurred on dead filaments of *Bulbochaete* sp. in Keston Ponds, Kent.

Chytridium cocconeidis Canter

Canter (1947*a**): on *Cocconeis pediculus* Ehrenb., Clissold Park Lake, London.

Chytridium confervae (Wille) Minden

Sparrow (1943, p. 341): on *Tribonema bombycina* (Ag.) Derb. et Sol., coll. Odam. Communication with Dr Odam gives the locality as Lower Sydenham, Kent.

Chytridium inflatum Sparrow

Sparrow (1936*): on *Oedogonium* sp., from site 5, Surrey (only empty sporangia found).

Chytridium lagenaria Schenk *pro parte* (=*Rhizidium westii* Massee=*Phlyctochytrium westii* (Massee) de Wildeman)

Massee (1891,* as *Rhizidium westii*): gregarious on *Spirogyra nitida* (Dillw.) Link and *Cladophora glomerata* (L.) Kütz., locality not given. Massee & Crossland (1905 as *Rhizidium westii*): on *Spirogyra nitida*, Frizinghall, Bradford, south-west Yorks (coll. W. West). Ramsbottom (1916*a*, *b*, as *P. westii*). Mason (1927, as *Phlyctochytrium westii*), south-west Yorks. Mason & Grainger (1947, as *P. westii*), vice-county 63, Yorks. Sparrow

(1936*): on *Rhizoclonium hieroglyphicum* Kütz., Coe Fen, Cambridge. Canter (unpublished): on zygospores of *Spirogyra* sp.; Chelsea Physic Garden pond, London and dead cells of *Cladophora* sp.; Clissold Park Lake, London.

Ramsbottom, Mason and Mason & Grainger cite this species as *Phlyctochytrium westii* (Massee) Lemmermann. Lemmermann (1901) transferred *Rhizidium westii* to *Phlyctochytrium westii* but this had already been done by de Wildeman (1896*a*) who therefore takes precedence.

Rhizidium westii, as described by Massee (1891), resembles *Chytridium lagenaria* but no operculum was observed, see Sparrow (1943, p. 349).

Chytridium lecythii (Ingold) Goldie-Smith (≡*Rhizophidium lecythii* Ingold)

Ingold (1941,* as *R. lecythii*) and Goldie-Smith (1946*): on *Lecythium hyalinum*, a rhizopod, Cropston Reservoir, Leicestershire.

Chytridium oedogonii Couch

Canter (1950*d**): saprophytic on *Oedogonium* sp., from the lake shore, Windermere (north basin), Blelham Tarn, Esthwaite Water, Derwentwater and Loweswater.

Chytridium olla Braun

Sparrow (1936*): on oogonia of *Nitella tenuissima* (?), Wicken Fen, Cambs.

Chytridium schenkii (Dang.) Scherffel

Sparrow (1936*): on *Oedogonium* sp., from site 5, Surrey. Sparrow (1943, p. 343) suggests that this fungus seems closer to *C. scherffelii* Sparrow.

Chytridium sphaerocarpum Dangeard

Sparrow (1936*): on *Mougeotia* sp., Coe Fen, Cambridge; on filaments of *Achlya* sp.; Barton Mills, Suffolk; and on *Spirogyra* sp., from site 1, Cambridge. Sparrow (1936, p. 438) suggests that a fungus on *Oscillatoria* sp., from site 1, Cambridge may belong to this species.

Chytridium versatile Scherffel

Sparrow (1936*): on *Synedra* sp., brook in St Andrew's Street, and Coe Fen, Cambridge. Canter (1950*c**) on: *Fragilaria crotonensis* (A. M. Edw.) Kitton, and on †*Tabellaria fenestrata* (Lyngb.) Kütz. var. *asterionelloides* Grün., Windermere (north and south basins): and on *Fragilaria crotonensis* in Esthwaite Water. Canter (unpublished): on *F. crotonensis*, Ullswater. Sparrow (1936, p. 437) states that the Coe Fen material resembles the incompletely known *Rhizophydium septocarpoides* Petersen. Sparrow (1936,* p. 437) also suggests that certain fungi found on *Tabellaria* sp., site 5, Surrey, may belong to this species.

† The name of this diatom according to Knudson (1952) is *Tabellaria flocculosa* (Roth) Kütz. var. *asterionelloides* (Grün.) Knudson.

Chytridium versatile Scherffel var. **aculis** Canter

Canter (1947*a**): on *Nitzschia sigmoidea* (Ehrenb.) W. Sm., in Bradbourne Park Lake, Sevenoaks, Kent, and from River Yeo, Sherborne, Dorset. I now suggest that the erection of this variety is probably superfluous. It seems that the presence or absence of an extramatrical stalk may be related to the presence or absence of mucilage around the wall of the diatom.

Chytridium xylophilum Cornu

Sparrow (1943, p. 359) discusses under this binomial a fungus he described (1936,* p. 432) as *Chytridium* sp. (n.g. ?): saprophytic on very old twigs of *Aesculus*, from site 2, Cambridge, and on twigs of *Quercus* sp., Kew Gardens, Surrey.

Zygorhizidium melosirae Canter

Canter (1950*c**): on *Melosira italica* (Ehrenb.) Kütz [now identified as *Melosira italica* subsp. *subarctica* O. Müll. by Dr J. W. G. Lund]; Esthwaite Water, Blelham Tarn, Windermere (north and south basins), Ullswater, Haweswater and Loughrigg Tarn. Canter (1951): Loweswater and Lough Derg (Galway and Clare). Canter (unpublished): Loch Ness (coll. Father A. McKillop).

Zygorhizidium parvum Canter

Canter (1950*c**): on *Sphaerocystis schroeteri* Chodat, Blelham Tarn, Esthwaite Water and Windermere (north and south basins). Canter (1951): Loweswater, Haweswater, Wastwater, Ullswater, Brothers Water, Swithland Reservoir and Lough Currane. Canter (unpublished): Bassenthwaite Lake, Malham Tarn, Loughs Talt and Oughter (coll. H. C. Gilson), Rostherne Mere (coll. E. M. Lind). Canter (1950*c*): on *Kirchneriella obesa* (W. West) Schmidle; Blelham Tarn; Canter (1951): on *Kirchneriella* sp., Newton Mere and Rhosneigr Lake.

Zygorhizidium willei Löwenthal

Canter (1947*b**): on *Mougeotia* sp., Montreal Park Lake, Sevenoaks, Kent. I have commonly found this fungus on *Mougeotia* sp., *Spirogyra* sp., *Cylindrocystis crassa* de Bary and *C. brebissonii* Menegh., in localities near Wray Castle.

Chytriomyces nodulatus Haskins

Haskins (1946*): saprophytic in leaves of wheat, maize, oats, various grasses and in insect exuviae submerged in water, Cambridge.

Chytriomyces tabellariae (Schroeter) Canter

Canter (1949*a**): on *Tabellaria flocculosa* (Roth) Kütz., Blelham Tarn bog near Wray Castle, and *T. fenestrata* (Lyngbye) Kütz. (= *Tabellaria* sp., see Canter, 1951, p. 150), Bassenthwaite Lake, Buttermere and Crummock Water. Canter (1951*): on *Tabellaria* sp., Derwentwater, Thirlmere and

Haweswater; on *T. fenestrata* var. *intermedia* Grün., Elterwater; on *T. flocculosa* (Roth) Kütz., Coniston Water, Brothers Water, Ullswater and Moss Eccles Tarn. [The name of the diatom for all these records according to Knudson (1952) is *Tabellaria flocculosa* (Roth) Kütz. var. *flocculosa* (Roth) Knudson.]

Nowakowskiella elegans (Nowak.) Schroeter

Sparrow (1936*): saprophytic on leaves of *Elodea*, Wicken Fen, Cambs, from site 2, Cambridge: on decayed grass leaves, ditch, rear of Queen's College, Cambridge.

Nowakowskiella hemisphaerospora Shanor

Haskins (1946): saprophytic on grass and regenerated cellulose film, Cambridge.

Amphicypellus elegans Ingold

Ingold (1944*): saprophytic on *Ceratium hirundinella* O.F.M. and *Peridinium* sp., Windermere. Canter (1951): saprophytic on *C. hirundinella*, Esthwaite Water, Blelham Tarn, Ullswater, Thirlmere, Loughrigg Tarn, Loweswater, Rostherne Mere, Knutsford Mere, and Loch Lomond. Canter (unpublished): Bassenthwaite Lake, Haweswater, Derwentwater and Loughs Talt, Arrow, and Glencar (coll. H. C. Gilson). Canter (1951): saprophytic on *Peridinium cinctum* Ehrenb., Blelham Tarn, Esthwaite Water, Loweswater, Thirlmere and Windermere (north and south basins). Canter (unpublished): Buttermere and Loughrigg Tarn.

EXCLUDED SPECIES

CATENARIA ANGUILLULAE Sorokin

Butler & Buckley (1927), Butler (1928), Buckley & Clapham (1929), and Butler & Humphries (1932). The genus *Catenaria* was transferred from the Chytridiales to the Blastocladiales by Couch (1945).

OLPIDIUM GILLII de Wildeman

Gill (1893*): in *Pleurosigma attenuatum* (Kütz.) W. Sm., *Cocconema lanceolatum* Ehrenb., *Nitzschia sigmoidea* (Ehrenb.) W. Sm., and *N.* sp., from the New River, London. Ramsbottom (1916*a*), Smith & Ramsbottom (1916). This fungus was originally described by Gill as *Ectrogella bacillariacearum* Zopf. It was suggested by de Wildeman (1896*b*) that Gill's figs. 1–8 represent a species of *Olpidium* for which he erected *O. gillii*. Friedmann (1952) identifies the fungus in *Pleurosigma attenuatum* with *Olpidiopsis gillii* (de Wildeman) Friedmann (Lagenidiales). It is stated, however, that the fungus in *Cymbella lanceolata* (*Cocconema lanceolatum*) and *Nitzschia sigmoidea* ought not to be included in *Olpidiopsis gillii.*

OLPIDIUM LEMNAE (Fisch) Schroeter (=*Reessia amoeboides* Fisch)

Included in Massee (1891) as two separate fungi *O. lemnae* Schroet., and *Reessia amoeboides* Fisch). Ramsbottom (1916*a*, *b*). It is thought that this fungus may be related to the Hyphochytriaceae.

OLPIDIUM SPHACELLARUM (Kny) Fischer (≡*Chytridium* (*Olpidium*) *sphacellarum* Kny)

Kny (1871 as *C.* (*O*). *sphacellarum*), Murray (1893, and extract of this paper by Holmes (1893), as *C. sphacellarum*). Johnson (1909), Ramsbottom (1916*a*), Smith & Ramsbottom (1916). Sparrow (1943, p. 629) places this fungus in the Olpidiopsidaceae as (?) *Olpidiopsis sphacellarum* (Kny) Sparrow.

OLPIDIUM TUMEFACIENS (Magnus) Berl. et de Toni (≡*Chytridium* (*Olpidium*) *tumefaciens* Magnus)

Magnus (1872, 1875), Murray (1893 and extract of this paper by Holmes (1893) and Wright (1879*a*) as *C. tumefaciens*). Ramsbottom (1916*a*), Smith & Ramsbottom (1916). Karling (1942, p. 26) and Sparrow (1943, p. 533) recognize this species as *Eurychasmidium tumefaciens* (Magnus) Sparrow (Ectrogellaceae).

RHIZOPHIDIUM DICKSONII Wright

Wright (1879*b*), Rattray (1887), Smith & Ramsbottom (1916). Karling (1942, p. 23) and Sparrow (1943, p. 528) recognize this species as *Eurychasma dicksonii* (Wright) Magnus (Ectrogellaceae).

LAKES AND SMALLER BODIES OF WATER CONTAINING CHYTRIDS OCCURRING ON PLANKTONIC ALGAE

ENGLAND
The Lake District
Bassenthwaite Lake
Blelham Tarn
Brothers Water
Buttermere
Coniston Water
Crummock Water
Derwentwater
Elterwater
Esthwaite Water
Haweswater
Loughrigg Tarn
Loweswater
Moss Eccles Tarn
Sunbiggin Tarn
Thirlmere
Ullswater
Wastwater
Windermere

Cheshire
Knutsford Mere
Newton Mere
Norton Mere
Rostherne Mere

Leicestershire
Swithland Reservoir

London
Barn Elms Reservoir 5
King George (Lea) Reservoir
Walthamstow Racecourse Reservoir

Shropshire
Hatch Mere

Yorkshire
Malham Tarn

NORTHERN IRELAND
Fermanagh
Lough Erne

EIRE
Cavan
Lough Oughter

Galway and Clare
Lough Derg

Kerry
Lough Currane

Leitrim
Lough Glencar

Sligo
Lough Arrow
Lough Talt

SCOTLAND
Dumbarton and Stirling
Loch Lomond

Inverness
Loch Mhor
Loch Ness

WALES
Anglesey
Llyn Coron
Rhosneigr Lake

INDEX TO SITE NUMBERS IN SPARROW (1936)

Site 1. Small stream in Chapman's Garden, Emmanuel College, Cambridge.
Site 2. Ditch to the rear of Jesus College, bordering on Midsummer Common, Cambridge.
Site 3. Ditch to the rear of Peterhouse, bordering Coe Fen, Cambridge.
Site 4. Ditch on the Newnham–Grantchester road by Trinity College playing field.
Site 5. The pond to the rear of the Educational Museum, Haslemere, Surrey.

PRECISE POSITION OF CERTAIN LOCALITIES NOT FULLY QUOTED IN THE TEXT

Chelsea Physic Garden, Chelsea, S.W. London.
Clissold Park Lake, Stoke Newington, N. London.
Rusland Moss, near Newby Bridge, N. Lancashire
Wray Castle, Low Wray, Claife, N. Lancashire.

LIST OF SUBSTRATA

(PLANTS)

ALGAE

CHLOROPHYCEAE
Volvocales
Apiocystis brauniana: Rhizophidium anomalum.
Chlamydomonas intermedia: Rhizophidium acuforme (?).*
Chlamydomonas sp.: Phlyctochytrium proliferum (?), Scherffeliomyces appendiculatus.
Chlorogonium elongatum: (?) Phlyctidium chlorogonii.
Eudorina elegans: Dangeardia mammillata, Endocoenobium eudorinae, Rhizophidium eudorinae, Rhizophidium transversum (?).
Gemellicystis neglecta: Rhizidium windermerense, Rhizophidium fulgens.
Sphaerocystis schroeteri: Rhizophidium sphaerocystidis, Zygorhizidium parvum.
Stylosphaeridium stipitatum: Rhizophidium ephippium.

Chlorococcales
Chlorococcum sp.?: Rhizophidium simplex.
Dictyosphaerium pulchellum: Rhizophidium sphaerocystidis.
Eremosphaera viridis: (?) Olpidium endogenum, (?) Rhizophidium ampullaceum.
Kirchneriella obesa: Zygorhizidium parvum.
Unicellular (spherical) green alga: (?) Rhizophidium sp.

---

* A (?) refers to fungi whose identification may be doubtful. Excluded species are omitted.

Ulothricales

Ulothrix sp.: Rhizophidium globosum, Rhizophidium laterale (?).

Oedogoniales

Bulbochaete sp.: Chytridium chaetophilum, (?) Entophlyctis bulligera.

Oedogonium crassusculum var. *idiosporum:* Rhizophidium ampullaceum.

Oedogonium rothii (oogonia): Chytridium acuminatum.

Oedogonium sp.: Chytridium inflatum, C. oedogonii, C. schenkii (?), Entophlyctis confervae-glomerata.

Oedogonium sporelings: Rhizophidium sp.

Cladophorales

Cladophora glomerata: (?) Chytridium lagenaria.

Cladophora sp.: Chytridium lagenaria, Entophlyctis confervae-glomerata, Olpidium entophytum, Phlyctochytrium quadricorne.

Rhizoclonium hieroglyphicum: Chytridium lagenaria.

Siphonales

Vaucheria sp.: Entophlyctis helioformis, E. rhizina, (?) Olpidium endogenum.

Conjugales

Closterium costatum: Micromycopsis mirabilis, Olpidium utriculiforme, Phlyctochytrium mucronatum.

Closterium dianae: Micromycopsis mirabilis, Olpidium utriculiforme.

Closterium kutzingii: Micromycopsis mirabilis.

Closterium lunula: Micromycopsis mirabilis, (?) Olpidium endogenum, O. utriculiforme.

Closterium pritchardianum: Phlyctochytrium mucronatum.

Cylindrocystis brebissonii: Endodesmidium formosum, Zygorhizidium willei.

Cylindrocystis crassa: Endodesmidium formosum, Zygorhizidium willei.

Hyalotheca dissiliens: Olpidium hyalothecae.

Mougeotia sp.: Chytridium sphaerocarpum, Micromyces laevis, M. petersenii, M. zygogonii, Olpidium endogenum (?), Phlyctidium apophysatum, Rhizophidium ampullaceum, Zygorhizidium willei.

Netrium oblongum: Endodesmidium formosum.

Spirogyra nitida: (?) Chytridium lagenaria.

Spirogyra sp.: Chytridium sphaerocaprum, Entophlyctis confervae-glomerata, (?) Entophlyctis sp., Micromyces zygogonii, (?) Olpidium endogenum, (?) Phlyctochytrium biporosum, P. laterale, Rhizidium variabile, R. simplex, (?) Rhizophidium spp., Zygorhizidium willei.

Spirogyra (zygospore): Chytridium lagenaria.

Spirotaenia condensata: Rhizophidium columnaris.

Staurastrum paradoxum: (?) Rhizophidium sp.

Tetmemorus brebissonii: Micromycopsis fischeri.

Zygnema sp.: Micromycopsis intermedia, Olpidium endogenum (?), (?) Pleotrachelus petersenii, (?) Rhizophidium barkerianum.

Charales

Chara sp.: Diplophlyctis intestina, D. laevis, Entophlyctis helioformis.

Nitella tenuissima ?: (?) Rhizophidium sp.

Nitella tenuissima (?) *(oogonia):* Chytridium olla.

Nitella sp.: Diplophlyctis intestina, Entophlyctis helioformis.

EUGLENINEAE

Euglena viridis: Polyphagus euglenae, P. laevis.

Euglena sp.: (?) Olpidium euglenae, Polyphagus euglenae (?), P. laevis.

Euglena (resting cells): (?) Entophlyctis apiculata, Polyphagus euglenae, P. laevis, Scherffeliomyces parasitans.

DINOFLAGELLATAE

Ceratium hirundinella: Amphicypellus elegans.

Peridinium cinctum: Amphicypellus elegans.

CHRYSOPHYCEAE

Hyalobryon mucicola (?) now (*H. polymorphum* Lund)*:* Rhizophidium hyalobryonis.

XANTHOPHYCEAE

Tribonema bombycina: Chytridium confervae.
Tribonema bombycina forma *minor:* Rhizophidium goniosporum.

BACILARIOPHYCEAE

Asterionella formosa: Rhizophidium planktonicum.
Cocconeis pediculus: Chytridium cocconeidis.
Fragilaria crotonensis: Chytridium versatile, Rhizophidium fragilariae.
Fragilaria sp.: Podochytrium clavatum.
Melosira italica subsp. *subarctica:* Zygorhizidium melosirae.
Melosira varians: Chytridium appressum, Podochytrium lanceolatum, Rhizidiopsis emmanuelensis, Rhizophidium fusus.
Melosira sp.: Rhizophidium fusus forma.
? Navicula sphaerophora: (?) Rhizophidium sp.
Nitzschia sigmoidea: Chytridium versatile var. aculis.
Nitzschia sp.?: Rhizidiopsis emmanuelensis.
Stauroneis phoenicenteron: (?) Rhizophidium sp.
Synedra sp.: Chytridium versatile, Rhizophidium fusus forma, Septolpidium lineare.
Tabellaria flocculosa var. *asterionelloides:* Chytridium versatile.
Tabellaria flocculosa var. *flocculosa:* Chytriomyces tabellariae.
Tabellaria sp.: (?) Chytridium versatile, Chytriomyces tabellariae.

CYANOPHYCEAE

Anabaena affinis var. *intermedia:* Rhizosiphon anabaenae, R. crassum.
Anabaena circinalis?: Rhizosiphon crassum.
Anabaena spiroides: Rhizosiphon anabaenae, R. crassum.
Anabaena spiroides var. *crassa:* Rhizosiphon crassum.
Anabaena sp.: Rhizosiphon crassum.
Oscillatoria aghardii var. *isothrix:* Rhizophidium megarrhizum.
Oscillatoria sp.: (?) Chytridium sphaerocarpum, Rhizophidium megarrhizum, R. subangulosum.

MARINE ALGAE

Callithamnion [*Antithamnion*] *plumulae:* (?) Olpidium plumulae.

FUNGI

Achlya spp.: Chytridium sphaerocarpum, Rhizophidium carpophilum, (?) Rozella septigena.
Chytridium oedogonii: Rozella sp.
Chytriomyces tabellariae: Septosperma anomala.
Dictyuchus monosporus: Rhizophidium carpophilum.
Micromycopsis sp. (sori): Septosperma anomala.
Monoblepharis macrandra: Rhizophidium carpophilum.
Polyphagus euglenae?: Rozella polyphagi.
Polyphagus laevis?: Rozella polyphagi.
Pythium intermedium: Rozella cuculus.
Pythium vexans ?: Rozella irregularis.
Pythium sp.: (?) Rozella sp.
Rhizophidium planktonicum agg.: Septosperma anomala.
Rhizophidium sphaerocystidis: Septosperma anomala.
Saprolegnia spp.: (?) Rozella septigena.
Zygorhizidium willei: Septosperma anomala.

POLLEN AND OTHER PLANT MATERIAL

Typha pollen: Chytridium chaetophilum, Rhizophidium sphaerothecae.
Twigs, Aesculus and Quercus: (?) Chytridium xylophilum.
Elodea leaves: Cladochytrium replicatum, Nowakowskiella elegans.
Grass: Chytriomyces nodulatus, Cladochytrium hyalinum, Entophlyctis aurea, Nowakowskiella elegans, N. hemisphaerospora.
Maize, wheat and oats: Chytriomyces nodulatus, Entophlyctis aurea, Diplophlyctis laevis.
Maize leaf and stems: Diplophlyctis intestina.

Oat leaf: Diplophlyctis laevis.
Oat leaf and stems: Diplophlyctis intestina.
Vegetable debris: (?) Rhizidium spp.

EXUVIAE
Chironomidae: Rhizidium mycophilum, Rhizoclosmatium globosum.
Unidentified exuviae: Asterophlyctis sarcoptoides, Chytriomyces nodulatus, Rhizoclosmatium globosum, Siphonaria variabilis.

ANIMALS
Anguillula: Rhizophidium vermicola.
Dobothriocephalus latus: Rhizophidium carpophilum (?).
Lecythium hyalinum: Chytridium lecythii.
Liver fluke eggs: Rhizophidium zoophthorum.
Rotifer eggs: Olpidium gregarium.
Rotifer: Rhizophidium globosum (?).

REFERENCES

ARCHER, W. (1860). On the occurrence of zoospores in the family Desmidiaceae. *Quart. J. micr. Sci.* **8**, 215–239.

ARCHER, W. (1867). *Chytridium Barkerianum. Quart. J. micr. Sci.* (N.S.), **7**, 89–90.

BARTSCH, A. F. (1945). The significance of zygospore character in *Polyphagus euglenae. Mycologia,* **37**, 553–570.

BUCKLEY, J. J. C. & CLAPHAM, P. A. (1929). The invasion of Helminth eggs by chytridiacean fungi. *J. Helminth.* **7**, 1–14.

BUTLER, E. J. (1907). An account of the genus *Pythium* and some chytridiaceae. *Mem. Dep. Agric. India, Bot.* **1**, 1–160.

BUTLER, E. J. (1928). Morphology of the chytridiacean fungus, *Catenaria anguillulae,* in liver-fluke eggs. *Ann. Bot., Lond.,* **42**, 813–821.

BUTLER, J. B. & BUCKLEY, J. J. C. (1927). *Catenaria anguillulae* as a parasite of the ova of *Fasciola hepatica. Sci. Proc. R. Dublin Soc.* **18**, 497–512.

BUTLER, J. B. & HUMPHRIES, A. (1932). On the cultivation in artificial media of *Catenaria anguillulae,* a chytridiacean parasite of the ova of liver fluke, *Fasciola hepatica. Sci. Proc. R. Dublin Soc.* **20**, 301–324.

CANTER, H. M. (1946). Studies on British Chytrids. I. *Dangeardia mammillata* Schröder. *Trans. Brit. mycol. Soc.* **29**, 128–134.

CANTER, H. M. (1947*a*). Studies on British Chytrids. II. Some new monocentric chytrids. *Trans. Brit. mycol. Soc.* **31**, 94–105.

CANTER, H. M. (1947*b*). Studies on British Chytrids. III. *Zygorhizidium willei* Löwenthal and *Rhizophidium columnaris* n.sp. *Trans. Brit. mycol. Soc.* **31**, 128–135.

CANTER, H. M. (1949*a*). Studies on British Chytrids. IV. *Chytriomyces tabellariae* (Schröter) n.comb. parasitized by *Septosperma anomala* (Couch) Whiffen. *Trans. Brit. mycol. Soc.* **32**, 16–21.

CANTER, H. M. (1949*b*). Studies on British Chytrids. V. On *Olpidium hyalothecae* Scherffel and *Olpidium utriculiforme* Scherffel. *Trans. Brit. mycol. Soc.* **32**, 22–29.

CANTER, H. M. (1949*c*). Studies on British Chytrids. VI. Aquatic Synchytriaceae. *Trans. Brit. mycol. Soc.* **32**, 69–94.

CANTER, H. M. (1949*d*). Studies on British Chytrids. VII. On *Phlyctochytrium mucronatum* n.sp. *Trans. Brit. mycol. Soc.* **32**, 236–240.

CANTER, H. M. (1950*a*). Studies on British Chytrids. VIII. On *Rhizophidium anomalum* n.sp. *New Phytol.* **49**, 98–102.

CANTER, H. M. (1950*b*). Studies on British Chytrids. IX. *Anisolpidium stigeoclonii* (de Wildeman) n.comb. *Trans. Brit. mycol. Soc.* **33**, 335–344.

CANTER, H. M. (1950*c*). Fungal parasites of the phytoplankton. I. (Studies on British Chytrids. X.) *Ann. Bot., Lond.,* N.S., **14**, no. 54, 263–289.

CANTER, H. M. (1950*d*). Studies on British Chytrids. XI. *Chytridium oedogonii* Couch. *Trans. Brit. mycol. Soc.* **33**, 354–358.

CANTER, H. M. (1951). Fungal parasites of the phytoplankton. II. (Studies on British Chytrids. XII.) *Ann. Bot., Lond.,* N.S., **15**, no. 58, 129–156.

Canter, H. M. & Lund, J. W. G. (1948). Studies on plankton parasites. I. Fluctuations in the numbers of *Asterionella formosa* Hass., in relation to fungal epidemics. *New Phytol.* **47**, 238–261.

Canter, H. M. & Lund, J. W. G. (1951). Studies on plankton parasites. III. Examples of the interaction between parasitism and other factors determining the growth of diatoms. *Ann. Bot., Lond.*, N.S., **15**, no. 59, 359–371.

Cook, W. R. I. (1932). An account of some uncommon British species of the chytridiales found in algae. *New Phytol.* **31**, 133–144.

Cook, W. R. I. (1936). Fungi of Glamorgan (Phycomycetes). *Glamorgan-County History*, I, 213–214.

Cooke, M. A. (1882–84). *British Freshwater Algae.* **1**, 1–329, London: Williams and Norgate.

Couch, J. N. (1945). Observations on the genus *Catenaria. Mycologia*, **37**, 163–193.

Friedmann, I. (1952). Über neue und wenig bekannte auf Diatomeen parasitierende Phycomyceten. *Öst. bot. Z.* **99**, 173–219.

Fritsch, F. E. (1903). Two fungi, parasitic on species of *Tolypothrix. Ann. Bot., Lond.*, **17**, 649–664.

Gill, C. H. (1893). On an endophytic parasite of diatoms. *J. R. micr. Soc.* **1**, 1–4.

Goldie-Smith, E. K. (1946). *Chytridium lecythii* (Ingold) n.comb. *Trans. Brit. mycol. Soc.* **29**, 68–69.

Grenfell, J. G. (1894). Fungi parasitic on diatoms. *J. Quekett micr. Cl.* Ser. II, vol. V, **34**, 371.

Griffiths, B. M. (1925). Studies in the phytoplankton of the Lowland Waters of Great Britain, no. III. The phytoplankton of Shropshire, Cheshire, and Staffordshire. *J. Linn. Soc. (Bot.)*, **47**, 75–98.

Grove, W. B. (1917). *Rhizophidium acuforme* Fisch. *New Phytol.* **16**, 177–180.

Gwynne-Vaughan, H. & Barnes, B. (1937). *The Structure and Development of the Fungi.* Cambridge University Press.

Haskins, R. H. (1939). Cellulose as a substratum for saprophytic chytrids. *Amer. J. Bot.* **26**, 635–639.

Haskins, R. H. (1946). New chytridiaceous fungi from Cambridge. *Trans. Brit. mycol. Soc.* **29**, 135–140.

Henfrey, A. (1859). On *Chlorosphaera*, a new genus of unicellular freshwater algae. *Trans. micr. Soc. Lond.* (N.S.) **7**, 25–29.

Holmes, E. M. (1893). Extract of 'Parasites on Algae' by Murray 1893. *Grevillea*, no. 100, 103.

Hood, O. (1910). On *Rhizophidium Eudorinae*, a new chytridiaceous fungus. *Proc. Bgham. nat. Hist. Soc.* **12**, 38–45.

Ingold, C. T. (1940). *Endocoenobium Eudorinae*, gen. et sp. nov., a chytridiaceous fungus parasitizing *Eudorina elegans* Ehrenb. *New Phytol.* **39**, 97–103.

Ingold, C. T. (1941). Studies on British Chytrids. I. *Phlyctochytrium proliferum* sp.nov. and *Rhizophidium lecythii* sp.nov. *Trans. Brit. mycol. Soc.* **25**, 41–48.

Ingold, C. T. (1944). Studies on British Chytrids. II. A new chytrid on *Ceratium* and *Peridinium. Trans. Brit. mycol. Soc.* **27**, 93–96.

Ingold, C. T. (1949). On the genus *Resticularia. Ann. Bot., Lond.*, N.S., **13**, 435–443.

Johnson, T. (1909). *Chrysophlyctis endobiotica* Schilb. (Potato-wart or Black Scab), and other Chytridiaceae. *Sci. Proc. R. Dublin Soc.* **12**, N.S., 131–144.

Karling, J. S. (1942). *Simple Holocarpic Biflagellate Phycomycetes.* New York City, U.S.A.

Karop, G. C. (1889). Extract from a monograph by Dr Zopf, 'Zur Kenntniss der Infections Krankheiten niederer Thiere u. Pflanzen'. *J. Quekett micr. Cl.*, Ser. II, **4**, 34–37.

Knudson, B. M. (1952). The diatom genus *Tabellaria*. I. Taxonomy and morphology. *Ann. Bot., Lond.*, N.S., **26**, no. 63, 421–440.

Kny, L. (1871). *Chytridium (Olpidium) sphacellarum. S.B. Ges. naturf. Fr. Berl.* pp. 93–97. (Also *Hedwigia*, **11**, 81–87, 1872.)

Lemmermann, E. (1901). Die parasitischen und saprophytischen Pilze der Algen. *Abh. naturw. Ver. Bremen*, **17**, 185–201.

Magnus, P. (1872). Ueber ein *Chytridium. S.B. Ges. naturf. Fr. Berl.* pp. 87–90.

Magnus, P. (1875). Die botanischen Ergebnisse der Nordseefahrt von 21 Juli bis 9 Sept. 1872. *Wiss. Meeresuntersuch. Abt. Kiel*, **2–3**, 59–80.

Mason, F. A. (1927). Yorkshire Phycomycetes. *Naturalist, Lond.*, pp. 267–274.

Mason, F. A. & Grainger, J. (1947). *A Catalogue of Yorkshire Fungi.* London.

Massee, G. (1891). *British Fungi. Phycomycetes and Ustilagineae.* London.

Massee, G. & Crossland, C. (1905). The fungus flora of Yorkshire (Botanical). *Trans. Yorks. Nat. Un.* **4**, 1–396 (1902–)1905.

Murray, G. (1893). Parasites on Algae. *Natural Science*, **2**, 120–123.

Ramsbottom, J. (1916*a*). A list of the British species of Phycomycetes, etc., with a key to the genera. *Trans. Brit. mycol. Soc.* **5**, 304–317.

Ramsbottom, J. (1916*b*). Notes on the list of British Phycomycetes. *Trans. Brit. mycol. Soc.* **5**, 318–323.

Rattray, J. (1887). Note on *Ectocarpus. Trans. roy. Soc. Edinb.* **32**, 589–600, 1884–5.

Reynolds, N. (1940). Seasonal variations in *Staurastrum paradoxum* Meyen. *New Phytol.* **39**, 86–89.

Richards, M. (1951). The life history of *Diplophlyctis laevis. Trans. Brit. mycol. Soc.* **34**, 483–488.

Scourfield, D. J. (1936). The occurrence of a minute fungus on a free swimming alga (a chytrid on *Chlorogonium*) in an Epping Forest pond. *Essex Nat.* **25**, 120–123.

Shadbolt, G. (1852). On the sporangia of some filamentous freshwater algae. *Trans. micr. Soc. Lond.* **3**, 168–170.

Skvortzow, B. W. (1925). Zur Kenntnis der Phycomyceten aus der Nordmandschurei, China. *Arch. Protistenk.* **51**, 428–433.

Smith, A. L. & Ramsbottom, J. (1916). New or rare microfungi. *Trans. Brit. mycol. Soc.* **5**, 231–248.

Smith, W. (1853). On the stellate bodies occurring in the cells of freshwater algae. *Quart. J. micr. Sci.*, N.S., **1**, 68–72.

Sparrow, F. K. (1933). New chytridiaceous fungi. *Trans. Brit. mycol. Soc.* **18**, 215–217.

Sparrow, F. K. (1936). A contribution to our knowledge of the aquatic Phycomycetes of Great Britain. *J. Linn. Soc.* (Bot.), **50**, 417–478.

Sparrow, F. K. (1937). Some chytridiaceous inhabitants of submerged insect exuviae. *Proc. Amer. phil. Soc.* **78**, 23–53.

Sparrow, F. K. (1943). *Aquatic Phycomycetes.* Ann. Arbor, U.S.A.

Thwaites, G. H. K. (1846–7). On the occurrence of tetraspores in algae. *Ann. Mag. nat. Hist.* **17**, 262–263.

Wager, H. (1913). Life-history and cytology of *Polyphagus Euglenae. Ann. Bot., Lond.*, **27**, 173–202.

Whiffen, A. J. (1944). A discussion of taxonomic criteria in the Chytridiales. *Farlowia*, **1**, 583–597.

Wildeman, E. de (1896*a*). Census Chytridinaearum. *Bull. Soc. Bot. Belg.* (*Mém.*), **35**, 7–69.

Wildeman, E. de (1896*b*). Notes mycologiques. VII. *Ann. Soc. belge micr.* (*Mém.*), **20**, 21–64.

Wright, E. P. (1879*a*). On a new species of parasitic green alga belonging to the genus *Chlorochytrium* of Cohn. *Trans. R. Irish Acad.* (*Sci.*), **26**, 355–368, 1877.

Wright, E. P. (1879*b*). On a species of *Rhizophydium* parasitic on species of *Ectocarpus*, with notes on the fructification of the *Ectocarpi. Trans. R. Irish Acad.* (*Sci.*), **26**, 369–380.

Zopf, W. (1884). Zur Kenntniss der Phycomyceten. I. Zur Morphologie und Biologie der Ancylisteen und Chytridiaceen. *Nova Acta Leop. Carol.* **47**, 143–236.

(*Accepted for publication* 3 *October* 1952)

INDEX TO GENERA AND SPECIES

[Synonyms are printed in *italics*]

acuforme (Zopf) Fischer, Rhizophidium, 283, 285
acuminatum Braun, Chytridium, 291
amoeboides Fisch, *Reessia*, 294
Amphicypellus Ingold, 294
ampullaceum (Braun) Rabenhorst, *Olpidium*, 283
ampullaceum (Braun) Fischer, Rhizophidium, 283
anabaenae (Rodhe et Skuja) Canter, Rhizosiphon, 287
anguilluae Sorokin, Catenaria, 294
anomala (Couch) Whiffen, Septosperma, 288
anomalum Canter, Rhizophidium, 283
apophysatum Canter, Phlyctidium, 282
apiculata (Braun) Fischer, Entophlyctis, 288
appendiculatum, Zopf, *Rhizidium*, 287
appendiculatus (Zopf) Sparrow, Scherffeliomyces, 287
appressum Sparrow, Chytridium, 291
Asterophlyctis Petersen, 290
'Asterospheres', 282
aurea Haskins, Entophlyctis, 288

bacillariacearum Zopf, Ectrogella, 278, 279, 294
barkerianum Archer, *Chytridium*, 283
barkerianum (Archer) Rabenhorst, Rhizophidium, 283
biporosum Couch, Phlyctochytrium, 286
boodlei Fritsch, Resticularia, 278
bulligera (Zopf) Fischer, Entophlyctis, 288

carpophilum (Zopf) Fischer, Rhizophidium, 283
Catenaria Sorokin, 294
chaetophilum Scherffel, Chytridium, 291
chlorogonii Serbinow, Phlyctidium, 282, 286
Chytridium Braun, 286, 291–3
Chytriomyces Karling, 293
cienkowskiana Zopf, Entophlyctis, 288, 289
Cladochytrium Nowakowski pro parte, 291
clavatum Pfitzer, Podochytrium, 287
cocconeidis Canter, Chytridium, 291
columnaris Canter, Rhizophidium, 283
confervae (Wille) Minden, Chytridium, 291
confervae-glomerata (Cienkow.) Sparrow, Entophlyctis, 288
corynephorus Zopf, *Septocarpus*, 286
crassum Scherffel, Rhizosiphon, 287
cuculus Butler, *Pleolpidium*, 281
cuculus (Butler) Sparrow, Rozella, 281

Dangeardia Schröder, 287
dicksonii (Wright) Magnus, Eurychasma, 295
dicksonii Wright, *Rhizophidium*, 295
Diplophlyctis Schroeter, 289

Ectrogella Zopf, 278, 279, 294
elegans Ingold, Amphicypellus, 294
elegans (Nowak.) Schroeter, Nowakowskiella, 294
emmanuelensis Sparrow, Rhizidiopsis, 287
Endochytrium, Sparrow, 289
Endocoenobium, Ingold, 290
Endodesmidium, Canter, 282
endogenum (Braun) Schroeter, Olpidium, 280
Entophlyctis Fischer, 288–9
Entophlyctis sp. Sparrow, 289
entophytum (Braun) Rabenhorst, Olpidium, 280
ephippium Canter, Rhizophidium, 283
eudorinae Ingold, Endocoenobium, 290
eudorinae Hood, Rhizophidium, 284
euglenae Dangeard, Olpidium, 280
euglenae Nowakowski pro parte, *Polyphagus*, 290
euglenae Nowak. emend Bartsch, Polyphagus, 290
euglenae var. *minor* Nowakowski, *Polyphagus*, 290
Eurychasma Magnus, 295
Eurychasmidium, Sparrow, 280, 295

fischeri Scherffel, Micromycopsis, 282
formosum Canter, Endodesmidium, 282
fragilariae Canter, Rhizophidium, 284
fulgens Canter, Rhizophidium, 284
fusiforme Fischer, *Pseudolpidium*, 279
fusus (Zopf) Fischer, Rhizophidium, 284

gillii (de Wildeman) Friedmann, Olpidiopsis, 294
gillii de Wildeman, *Olpidium*, 294
globosum (Petersen) Sparrow, Rhizoclosmatium, 290
globosum (Braun) Rabenhorst, Rhizophidium, 284
goniosporum Scheffel, Rhizophidium, 284
gracile Zopf, Lagenidium, 279
gregarium (Nowak.) Schroeter, Olpidium, 280

haynaldii (Schaarschmidt) Fischer, Rhizophidium, 286
helioformis Dangeard, *Chytridium*, 288
helioformis (Dang.) Ramsbottom, Entophlyctis, 288
hemisphaerospora Shanor, Nowakowskiella, 294
hyalinum Berdan, Cladochytrium, 291
hyalobryonis Canter, Rhizophidium, 284
hyalothecae Scherffel, Olpidium, 280
'Hyperparasites', 288

inflatum Sparrow, Chytridium, 291
intermedia Canter, Micromycopsis, 282
intestina (Schenk) Schroeter, Diplophlyctis, 289
intestinum Schenk pro parte, *Rhizidium*, 289
irregulare Butler, *Pleolpidium*, 281
irregularis (Butler) Sparrow, Rozella, 281

laevis Sparrow, Diplophlyctis, 289
laevis Canter, Micromyces, 282
laevis (Nowak.) Bartsch, Polyphagus, 290
lagenaria Schenk pro parte, Chytridium, 291
Lagenidium Schenk, 279, 281
lagenula (Braun) Fischer, Rhizophidium, 284
lanceolatum Sparrow, Podochytrium, 287

laterale Sparrow, Phlyctochytrium, 286
laterale (Braun) Rabenhorst, Rhizophidium, 284
lecythii (Ingold) Goldie Smith, Chytridium, 292
lecythii Ingold, *Rhizophidium*, 292
lemnae (Fisch) Schroeter, Olpidium, 294
lineare Sparrow, Septolpidium, 281

mammillata Schröder, Dangeardia, 287
megarrhizum Sparrow, Rhizophidium, 284
melosirae Canter, Zygorhizidium, 293
Micromyces Dangeard, 278, 282
Micromycopsis Scherffel, 278, 282
mirabilis Canter, Micromycopsis, 282
mucronatum Canter, Phlyctochytrium, 286
mycophilum Braun, Rhizidium, 289
Myzocytium, Schenk, 279

nodosa Dangeard, Resticularia, 278
nodulatus Haskins, Chytriomyces, 293
Nowakowskiella Schroeter, 294

oedogonii Couch, Chytridium, 292
olla Braun, Chytridium, 292
Olpidiopsis, Cornu, 281, 294, 295
Olpidium (Braun) Rabenhorst, 279–80, 294, 295

parasitans Sparrow, *Scherffelia*, 287
parasitans (Sparrow) Sparrow, Scherffeliomyces, 287
parvum Canter, Zygorhizidium, 293
Petersenia Sparrow, 281
petersenii Scherffel, Micromyces, 282
petersenii Lund, Pleotrachelus, 281
Phlyctidium (Braun) Rabenhorst, 282
Phlyctochytrium Schroeter, 286–7
Physoderma Wallroth, 278
planktonicum Canter, Rhizophidium, 285
Pleolpidium Fischer, 278, 281
Pleotrachelus Zopf, 281
plumulae Cohn, *Chytridium*, 280
plumulae (Cohn) Fischer, Olpidium, 280
Podochytrium, Pfitzer, 286, 287
polycystis Cornu, Woronina, 278, 279
polyphagi Sparrow, *Pleolpidium*, 281
polyphagi Sparrow, *Pleolpidium* (Rozella), 281
polyphagi (Sparrow) Sparrow, Rozella, 281
Polyphagus Nowakowski, 290
proliferum Ingold, Phlyctochytrium, 286
Pseudolpidium Fischer, 279, 281
pythii (Butler) Karling, Olpidiopsis, 281
pythii Butler, *Pseudolpidium*, 281

quadricorne (de Bary) Schroeter, Phlyctochytrium, 287

rabenhorstii Zopf, Lagenidium, 279
Reessia Fisch, 294
replicatum Karling, Chadochytrium, 291
Resticularia Dangeard, 278
Rhizidiopsis Sparrow, 287
Rhizidium Braun, 289
Rhizidium spp., Sparrow, 289
rhizina (Schenk) Minden, Entophlyctis, 289
Rhizoclosmatium Petersen, 290
Rhizophidium Schenk, 283–6, 295
Rhizophidium sp., Canter, 286
Rhizophidium sp., Grenfell, 286
Rhizophidium sp., Reynolds, 286
Rhizophidium spp., Sparrow, 286
Rhizosiphon Scherffel, 287
rostellatum (de Wildeman) Fischer, Rhizophidium, 286
Rozella Cornu, 281
Rozella sp., Canter, 281
Rozella sp., Sparrow, 281

sarcoptoides Petersen, Asterophlyctis, 290
schenkii (Dangeard) Scherffel, Chytridium, 292
Scherffelia Sparrow, 287
scherffelii Sparrow, Chytridium, 292
Scherffeliomyces Sparrow, 287
septigena Cornu, Rozella, 281
septocarpoides Petersen, Rhizophidium, 292
Septocarpus Zopf, 286
Septolpidium Sparrow, 281
Septosperma Whiffen, 288
simplex (Dangeard) Fischer, Rhizophidium, 285
Siphonaria Petersen, 290
sphacellarum Kny, *Chytridium* (*Olpidium*), 295
sphacellarum (Kny), Sparrow, Olpidiopsis, 295
sphacellarum (Kny) Fischer, *Olpidium*, 295
sphaerocarpum Dangeard, Chytridium, 292
sphaerocarpum (Zopf) Fischer, Rhizophidium, 285
sphaerocystidis Canter, Rhizophidium, 285
sphaerothecae Zopf, Rhizophidium, 285
spirogyrae Skvortzow, Olpidium, 280
subangulosum (Braun) Rabenhorst, Rhizophidium, 285

tabellariae (Schroeter) Canter, Chytriomyces, 293
transversum (Braun) Rabenhorst, Rhizophidium, 285
tumefaciens Magnus, *Chytridium* (*Olpidium*), 295
tumefaciens (Magnus) Sparrow, Eurychasmidium, 280, 295
tumefaciens (Magnus) Berl. et de Toni, *Olpidium*, 295

Urophlyctis Schroeter, 278
utriculiforme Scherffel, Olpidium, 280

variabile Canter, Rhizidium, 289
variabilis Petersen, Siphonaria, 290
vermicola Sparrow, Rhizophidium, 285
versatile Scherffel, Chytridium, 292
versatile var. aculis Canter, Chytridium, 293

westii (Massee) de Wildeman, *Phlyctochytrium*, 291, 292
westii Massee, *Rhizidium*, 291, 292
willei Löwenthal, Zygorhizidium, 293
windermerense Canter, Rhizidium, 289
Woronina Cornu, 278, 279

xylophilum Cornu, Chytridium, 293

zoophthorum (Dangeard) Fischer, Rhizophidium, 286
zygogonii Dangeard, Micromyces, 282
Zygorhizidium Löwenthal, 293

THE SOIL FUNGI OF WICKEN FEN

By H. STENTON

Department of Botany, University College, Hull

(With 3 Text-figures)

The fungal flora of the alkaline, peaty soil of Wicken Fen, Cambridgeshire, was studied by the soil plate method for a period of two years. Approximately 120 species of fungi, including representatives of all the major classes, have been isolated. The commonest genera were *Mortierella*, *Pythium*, *Cylindrocarpon*, *Penicillium*, *Volutella*, *Trichoderma*, *Stysanus*, *Cephalosporium* and *Cladosporium*. The number of fungi isolated decreased with increasing depth and two groups of fungi could be distinguished by the position in the profile of their respective zones of greatest development. Few of the fungi were restricted in their horizontal distribution, but quantitative differences could be distinguished between the fungal floras of the two sampling sites. Several species of Basidiomycetes, including *Corticium coronilla* v. Höhnel & Litsch, were isolated regularly.

INTRODUCTION

Although the study of soil fungi began more than half a century ago (Waksman, 1944; Chesters, 1949), comparatively few investigations have so far been made of the fungal floras of undisturbed, natural soils. Wicken Fen was selected as the site of this investigation for the following reasons: (1) the apparent absence of any data on the fungi of natural, alkaline, peaty soils; (2) the detailed information already available concerning the ecology of the higher plants of this area; (3) the contrasting lack of information about the soil fungi of the area apart from the floristic list of higher fungi compiled by Corner (1934). The fen is situated in the parish of Wicken, Cambridgeshire, midway between Newmarket and Ely. It originally owed its preservation to its use as a 'Catch-water' into which the water draining into the fens from the higher land to the south and east was diverted. It presents a patchwork of vegetation of different types and all stages of development from open water to fen woodland or carr (Godwin, 1929). There has been no direct interference with the soil which is thus in, at least, a semi-natural state. The abundant water has retarded the decomposition of the dead plant remains and a considerable thickness of peat overlying gault clay covers the fen. Godwin, Mobbs & Barucha (1932) have investigated the pH and the soil atmosphere of the peat and find that, in general, the reaction is alkaline with a probably temporary tendency to slight acidity at the extreme surface. The pH values obtained colorimetrically varied from 6·5 to 8·0; very few were below pH 7·0.

The details of the soil profile are broadly the same throughout the fen, but the thickness of the zones varies from place to place. The surface zone, of some 4 in. in thickness, consists of a black or dark brown soil, pasty when wet and powdery when dry, and showing few coarse fragments of vegetable debris. Below this comes a zone of black peat about 8 in. thick, which overlies a reddish brown peat of variable depth. A layer of shell marl

2–6 in. thick extends over most of the fen, at a depth of 7–12 in. In the fen carr the first 6 in. or so of soil is powdery and friable, because much of it has been eaten by insect larvae and voided as faecal pellets. Godwin (1931) and Godwin & Barucha (1932) have investigated the water relations of the fen in great detail. The artificial control of the drainage by the sluice gates at Upware narrows the range of movement of the water-table. Nevertheless, the water-table is very near the surface in winter and much of the fen is actually flooded. The high porosity of the soil renders the water-table very sensitive to both rainfall and transpiration. This high water-table made the collection of a continuous series of profile samples from various parts of the fen very difficult. Many of the original sites became flooded in the course of the first winter and were abandoned. Eventually two sites were found where the water-table remained sufficiently low for an adequate portion of the soil profile to be sampled at intervals throughout the year. Site *A* was in a drove where the vegetation was similar to that of the Molinietum. The dominant plant was *Molinia coerulea*; whilst the following plants were also commonly present: *Phragmites communis*, *Filipendula (Spiraea) ulmaria*, *Galium palustre*, *Symphytum officinale*, *Polygala vulgaris*, *Thalictrum flavum* and *Carex panicea*. Site *F* was in a patch of old fen carr about two-thirds of a mile from site *A* with which it contrasted strongly. The soil samples were obtained beneath a large tree of *Rhamnus catharticus*. There was little surface vegetation, apart from a few stolons of *Rubus caesius* and a few plants of *Phragmites communis*. Near the edge of the canopy were some tussocks of *Deschampsia caespitosa*. The soil was covered with a litter of dead *Rhamnus* leaves, twigs and fruits. The surface soil was very friable and powdery. The water-table here was always much lower than at site *A*. It should be noted that even at these two sites the fluctuations in the level of the water-table influenced the depth to which the profile could be sampled.

METHODS

Soil samples for analysis were collected as follows: on each sampling occasion after scraping away the surface litter a small pit about 12 in. square was dug and the peat carefully removed. Sterile glass specimen tubes 2 × 1 in. were inserted into one vertical face of the pit, and a cylinder of soil removed in each. The first tube was inserted 1 in. below the surface, the next at 2 in. and the rest at 2 in. intervals. Since that portion of the contents of each tube which was used for the isolation of the fungi came from at least 1 in. behind the exposed face of the profile, the chance of contamination during collection was slight. The depth to which the profile could be sampled was controlled by the level of the water-table and, as previously mentioned, this varied from month to month and from site to site. At site *A* the greatest depth sampled varied from 8 to 30 in., the maximum depth usually being 16 in. At site *F* the greatest depth sampled varied from 15 to 33 in., the maximum depth usually being 24 in. Samples were collected at intervals of 3–4 weeks, and the distribution data presented later are based on eighteen such collections made between October 1948 and June 1950. The fungi were isolated by the soil-plate method of Warcup (1950). The isolation medium was Czapek-Dox agar containing 0·5 g.

Difco yeast extract and 30·0 g. sucrose and adjusted to pH 4·0 with phosphoric acid. Acidified starch-peptone agar was also used as a supplementary isolation medium, since *Mortierella* spp. fruited more readily on this weaker medium than on yeast-Dox agar. The baiting technique, in which boiled, split hemp seeds are added to a mixture of soil and water, was also used for the isolation of some of the Phycomycetes.

From each tube of soil three soil plates were prepared using yeast-Dox agar. In most cases two additional plates were prepared using starch-peptone agar. The plates were incubated at 22·5° C. and examined at intervals of 2–3 days, starting on the second day of incubation. Using a low-power, binocular microscope the whole plate could be examined fairly rapidly. As each plate was examined the fungi present were recorded against the depth of the sample. To obtain a positive record the fungi had to occur on one or more of the five plates. This allowed to some extent for the chance absence of a fungus from some of the plates and for its possible suppression by other fungi. Most of the fungi appeared on the plates during the first 2 weeks of incubation. Some appeared and fruited very early, e.g. *Mucor rammanianus* and *Mortierella isabellina*, and being low-growing forms, were easily overlooked if the examination of the plates was delayed. The plates were retained for a month because some slow-growing fungi appeared after the others had begun to die down. Some of the fungi could be recognized and recorded at once, but it was often necessary to isolate others and to induce them to fruit before they could be included in the list. In the examination of eighteen profiles from sites *A* and *F* approximately 2070 soil plates were prepared and about 35,000 colonies were examined. A representative culture of each fungus was kept and identified if possible. From this culture a slide and a herbarium specimen (prepared by drying a Petri-dish culture) were made and kept for future reference.

FUNGI ISOLATED FROM WICKEN FEN SOILS

Representatives of the four major classes of fungi were isolated from the soil at Wicken Fen. The approximate numbers of fungi in each class are as follows:

	Species		Species
Phycomycetes	12	Basidiomycetes	4
Ascomycetes	11	Fungi Imperfecti	91

Phycomycetes

(*a*) *Oomycetes*

Only two genera of Oomycetes were found. A species of *Saprolegnia* was isolated once only from a depth of 12 in. at site *A* using hemp seed bait. The genus *Pythium*, on the other hand, is very abundant in this soil, and is represented by at least two species. This genus is absent from the lists of soil fungi published by most earlier workers before Chesters (1948) and Warcup (1951*a*), probably because most of the earlier workers used the dilution plate method, which tends to favour freely sporing fungi, to the exclusion of *Pythium*. In this investigation *Pythium* was easily recognized and isolated from the soil plates. Indeed, it often proved to be a nuisance, suppressing other fungi by its vigorous growth. It was also isolated from hemp seed bait.

(*b*) *Zygomycetes*

The genus *Mortierella* proved to be the most abundant Phycomycete. *M. minutissima* was the most abundant species, and it occurred with great constancy and in great numbers at both sites. Bisby, Timonin & James (1935) state that *Mortierella* is the commonest Phycomycete in Manitoba soils. According to Warcup (1951*a*) it is the most abundant Phycomycete in Breckland soils. This genus also does not figure in the lists of earlier workers, probably because these fungi remain sterile on many of the common isolation media. The other common species at Wicken was *M. isabellina*. The genus *Mucor* was not well represented at Wicken. *M. silvaticus* and *M. rammanianus* were the only species which occurred regularly. Two forms of *M. rammanianus* were found. The commoner variety had intramatrical giant-cells, which the other lacked. *Absidia glauca* and *A. cylindrospora* were isolated but they were not very common. *Coemansia pectinata* appeared regularly at site *F* but only rarely at site *A*. One species of *Syncephalis* was commonly found on the hyphae of *Mortierella minutissima*. Attempts to bring it into culture were unsuccessful.

Ascomycetes

Eleven species of Ascomycetes were recorded. Of these an unidentified member of the Plectascales was the most important. This fungus, W38, occurred with great constancy in all soils examined. The next commonest Ascomycete was *Chaetomium cochloides*. *C. spinosum* occurred less frequently. An interesting find was the Discomycete, *Pyronema confluens*, which was isolated from soil taken at 11 in. below the surface at site *A*. It was isolated on starch-peptone agar and fruited on the soil plate and later in pure culture. Several ascosporic Aspergilli and Penicillia were found occasionally.

Basidiomycetes

Three or four species of Basidiomycetes were isolated regularly. The commonest species was *Corticium coronilla*.

Fungi imperfecti

The majority of the soil fungi of Wicken Fen belong to this class. Many of the species were recorded once only. The genera *Penicillium*, *Trichoderma*, *Cylindrocarpon*, *Volutella*, *Cladosporium*, *Cephalosporium*, *Verticillium*, *Paecilomyces*, *Gliomastix*, *Stysanus*, *Papularia*, *Gliocladium*, *Fusarium*, *Sporotrichum*, *Stachybotrys* and *Spicaria* were, however, constantly occurring members of the soil flora. Of the unidentified imperfect fungi, isolates W28, W35 and W133 represent fungi which occur regularly in the soil. Fungi which, on the isolation plates, produce colonies consisting of a mass of dark coloured, often sterile hyphae constitute an important component of the soil flora at Wicken. These 'dematiaceous' fungi were most abundant at site *F*. They were mostly sterile forms, and for this reason were not studied in detail. In addition to the dark coloured colonies several other types of sterile mycelium were found quite regularly.

List of species isolated

The following list (Table 1) includes approximately 120 fungi isolated from the soil at Wicken Fen. It is by no means complete, and there are certainly many more species to be isolated. A number of fungi found during the course of the investigation have not been included in the list because they could not be sufficiently well characterized. This was due to the failure to induce fruiting, or to the difficulty of correlating the taxonomic data obtained in cultural studies with those in the literature based on specimens growing on and in natural substrata. The figures denote the number of times each fungus was recorded during the analysis of eighteen complete profiles from both sites. Thus *Papularia arundinis* occurred in eleven profile samples from site *A* and in eight from site *F*. Those fungi which were recorded in nine or more of the eighteen samples from either site *A* or site *F* have been designated 'abundant' and their distribution in the soil has been examined in detail. Warcup (1952) states that in preparing a replicate series of soil plates, the amount of soil used in each plate was approximately the same (0·013 g.). Similar quantities of soil were used for the isolation of the Wicken fungi, so this figure (0·013 g.) will be taken to give an approximate idea of the implication of the term 'abundant' as used in the present paper. In sampling a profile an average of some eleven samples at increasing depths were taken, and from each sample, five sub-samples of the order of 0·013 g. were taken for the preparation of five soil plates. For a fungus to be present in any single profile, therefore, it must occur in any soil sample of size $11 \times 5 \times 0{\cdot}013 = 0{\cdot}7$ g. For a fungus to be designated 'abundant' in the sense used above, the probability of its occurrence in such a soil sample of approximately 0·7 g. must equal or exceed 50 % (i.e. occurrence in nine or more of eighteen complete profile samples). 'Occurrence' as used here implies both occurrence in the soil sample and ability to develop into a recognizable colony on one or more of the soil plates.

DISTRIBUTION OF THE 'ABUNDANT' FUNGI

Vertical distribution

The generalization that fungi are abundant in the surface layers of the soil, their numbers decreasing rapidly with depth, holds good at Wicken. The reason for this decrease in numbers is not obvious, but it is certainly not due to the scarcity of organic matter in the lower part of the profile, an explanation suggested for other types of soil. The fungi fall into two groups: (1) those most abundant near the surface of the soil; and (2) those most abundant in the subsurface zone (Table 1). The vertical distribution of the 'abundant' fungi was demonstrated by plotting the number of times they occurred in the samples against the depth at which the samples were taken. Figs. 1 and 2 are profile diagrams of typical members of each of the two groups. The horizontal, dotted line in each diagram indicates the level at which the interference of the water-table with sampling became so great as to render the validity of the distribution data rather doubtful, because the estimate of percentage occurrence (50 % occurrence and above = 'abundant') was based on fewer than eighteen samples.

Table 1. *List of fungi isolated*

		Occurrence (max. 18)	
	Phycomycetes	Site *A*	Site *F*
Oomycetes			
S AF	*Pythium* spp.	17	13
	Saprolegnia sp. (W 56)	1	0
Zygomycetes			
	Absidia glauca Hagem	3	5
	A. cylindrospora Hagem	0	1
S F	*Coemansia pectinata* Bainier	1	10
	Mortierella isabellina Oudemans	3	0
S AF	*M. minutissima* van Tiegh.	18	18
	Mucor rammanianus Moller	8	8
S F	*M. silvaticus* Hagem	6	14
	M. spinosus van Tiegh.	1	0
	Rhizopus nigricans Ehren.	1	0
S F	*Syncephalis* sp.	6	9
	Ascomycetes		
Plectomycetes			
	Aspergillus amstelodami (Mangin) Thom & Church	0	1
	A. fischeri Wehmer	0	2
	A. nidulans (Eidam) Wint.	0	1
	Aspergillus sp. (W 93)	0	1
	Penicillium wortmanni Klocker	0	1
	Perisporium vulgare Corda	0	1
SS AF	W 38	11	18
	W 40	0	2
Pyrenomycetes			
	Chaetomium cochloides Palliser	7	0
	C. spinosum Chivers	2	3
Discomycetes			
	Pyronema confluens Pers.	1	0
	Fungi imperfecti		
	Acrostalagmus cinnabarinus Corda	6	5
	Alternaria sp. (W 46)	4	3
	Alternaria sp. (W 124)	0	1
	Aspergillus sydowi (Bain. et Sart.) Thom & Church	0	1
	A. terreus Thom	0	3
	A. versicolor (Vuill.) Tiraboschi	6	8
	Aspergillus sp. (W 33)	0	1
	Botryosporium longibrachiatum Oudem.	0	1
	B. pulchrum Corda	1	0
	Botrytis cinerea Pers. ex Fr.	6	7
S AF	*Cephalosporium acremonium* Corda	10	14
	Cephalosporium sp. (W 74)	1	1
	Cephalosporium sp. (W 82)	1	0
S A	*Cephalosporium* sp. (W 83)	15	0
	Cephalosporium sp. (W 175)	0	1
	C. roseum Corda	1	1
S AF	*Cladosporium herbarum* Link	15	15
	Clonostachys pseudobotrytis ? (W 174) von Höhnel	1	0
	Coremium sp. (W 94)	1	0
S AF	*Cylindrocarpon radicicola* Wollenweber	18	18
S AF	*Dematiaceae* (several spp.)	18	18
S AF	*Fusarium culmorum* (W. G. Smith) Sacc.	10	12
	F. oxysporum ? sensu Snyder & Hansen	1	0
	Fusarium spp. W 44, W 84	1 each	0
	Fusarium sp. (W 142)	0	1
	Geomyces cretaceus Traaen	1	1

Table 1 (*continued*)

			Occurrence (max. 18)	
		Fungi imperfecti	Site *A*	Site *F*
S	*F*	*Gliocladium catenulatum* Gilman & Abbott	6	11
		G. roseum (Link) Bainier	3	0
S	*F*	*Gliomastix convoluta* var. *felina* (Marchal) Mason	7	9
		Hormiactis sp. ? (W 161)	1	0
		Myrothecium sp. (W 23)	2	5
		Myrothecium sp. (W 163)	1	0
		Nigrospora sp. (W 158)	2	0
		Oidiodendron sp. (W 135)	1	1
S	*AF*	*Paecilomyces* sp. (W 113)	17	17
		P. varioti Bainier	1	1
S	*A*	*Papularia arundinis* (Corda) Fr.	11	8
		Penicillium albidum Sopp	0	1
		P. claviforme Bainier	1	0
S	*F*	*P. expansum* Link	2	11
		P. frequentans Westling	1	0
		P. funiculosum Thom	3	0
		P. nigricans (Bain.) Thom	7	8
		P. phoeniceum van Beyma	0	1
		P. spinulosum Thom	1	0
S	*AF*	*P. thomii* Maire	13	9
S	*F*	*P. variabile* Sopp	2	15
		Penicillium spp. W 6*c*, W 11, W 100, W 102. W 106, W 107, W 137	0	1 each
		Penicillium spp. W 8, W 9, W 14*a*, W 16	1 each	0
		Penicillium sp. (W 14*b*)	2	0
		Penicillium sp. (W 36)	2	1
		Penicillium sp. (W 90)	0	3
		Periconia sp. (W 180)	1	0
		Pestalotia sp. (W 122)	1	2
		Rhinotrichum sp. (W 159)	1	0
		Scopulariopsis brevicaulis (Sacc.) Bainier	0	2
		Sepedonium sepedonioides Harz	1	0
S	*F*	*Spicaria violacea* Abbott	3	9
		Sporocybe sp. (W 181)	1	0
		Sporotrichum sp. (W 39)	1	3
S	*F*	*Sporotrichum* sp. (W 79)	4	9
S	*A*	*Stachybotrys atra* Corda	9	6
		Stemphylium sp. (W 87)	0	4
S	*F*	*Stysanus stemonites* (Pers.) Corda	8	10
S	*F*	*Stysanus* sp. (W 50*a*)	0	9
S	*AF*	*Trichoderma viride* Pers. ex Fr.	17	18
		Verticillium sp. (W 70)	0	2
S	*A*	*Verticillium* sp. (W 182)	14	0
S	*AF*	*Volutella ciliata* Fr.	18	18
S	*F*	W 28	0	12
S	*A*	W 35	9	6
		W 73	2	0
SS	*A*	W 133	11	0
		W 136, W 173	0	1 each
		W 147, W 160, W 171, W 172	1 each	0
		W 148	3	0
		W 162	0	2
		Mycelia sterilia		
		White mycelium, blackening with age (W 184)	5	3
SS	*F*	Brown, scented fungus (W 185)	8	9
		Coarse, orange mycelium	5	1

S = most abundant near surface; *SS* = most abundant in the subsurface zone; *A* = isolated in 50 % or more samples at site *A*; *F* = isolated in 50 % or more samples at site *F*; *AF* = isolated in 50 % or more samples at both sites *A* and *F*.

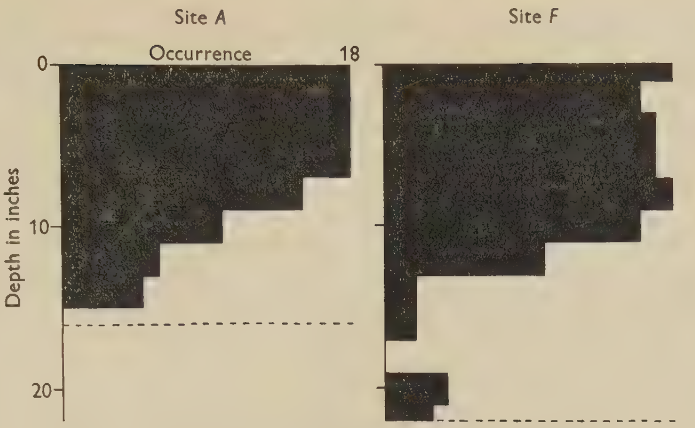


Fig. 1. Occurrence of *Mortierella minutissima* in Wicken soils.

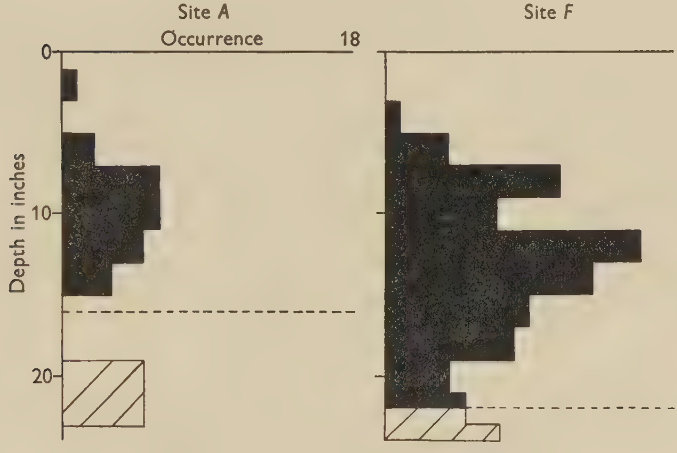


Fig. 2. Occurrence of W38 in Wicken soils.

Horizontal distribution

Few of the more abundant members of the fungal flora of the soil at Wicken are restricted in their distribution. Most of the 'abundant' fungi were isolated from both sites. However, two of these fungi were frequently isolated at site *A* and never at site *F*. These were *Cephalosporium* sp. (W83) and *Verticillium* sp. (W182). Similarly, there were two fungi which occurred regularly at site *F* but which were never isolated at site *A*, viz. W28 and *Stysanus* sp. (W50*a*). All the other abundant fungi were found at both of the sites. Nevertheless, they differed considerably in their abundance in the two soils. Thus of the thirty-one fungi studied in detail six were more abundant at site *A* than at site *F*, thirteen fungi were more abundant at site *F* than at site *A* and twelve fungi occurred in 50 % or more of the samples from both sites (Table 10). The differences in the fungal flora of the soil at sites *A* and *F* are thus quantitative rather than qualitative. Nevertheless, it is impossible to confuse a series of plates from site *A* with a series from site *F*. Those from site *A* are especially characterized by the colonies of *Cephalosporium* sp. (W83), which are pure white with a canary-yellow reverse and have a clearly defined outline. These, together with the brown colonies of *Cylindrocarpon radicicola* and the more diffuse white colonies of *Mortierella minutissima*, form a typical and easily recognized association. Soil plates from site *F* are characterized by colonies of *Penicillium expansum*, especially in samples from the upper part of the soil profile, and by the relative abundance of dematiaceous species. The large population of *P. expansum* at site *F* is probably correlated with the presence of numerous fruits of *Rhamus catharticus*. This fungus is often found on the fruits lying on the surface of the ground.

Direct examination of fungi in the soil

The fungi present in Wicken Fen soil were studied: (1) by mounting particles of soil in dilute lactophenol cotton-blue and examining under the microscope; (2) by staining soil plates with dilute lactophenol cotton blue after a short incubation period (Warcup, 1951 *a*, *b*). By the latter method the origin of the very young colonies could often be traced. Many such young colonies were found to have arisen from free spores or from spores embedded in humus particles. Much free mycelium was seen in soil from the upper layers of the profile, but most of it lacked stainable contents and was presumably dead. A smaller amount took up the stain and sometimes such mycelium had spores attached to it. In addition to the free mycelium, fragments of plant debris were often permeated by networks of hyphae. At depths greater than 5 in. the amount of mycelium and spores was much reduced. Occasionally large masses of apparently viable mycelium were seen.

Soil basidiomycetes

Until recently (Warcup, 1951 *c*), Basidiomycetes were rarely isolated from the soil. It is of interest to note that several species of these fungi were regularly isolated from the soils of Wicken Fen. Between June 1949 and June 1950, thirty-seven colonies bearing clamp connexions were observed on soil plates from eleven consecutive profile samples from sites *A* and *F*. These fungi appeared 10–14 days after the plates had been poured. From observations made on eleven of these isolates grown on 3 % malt agar under identical conditions, it appeared that at least three different fungi were represented. One isolate (B8) differed completely from the others in having characteristically beaded intramatrical hyphae but virtually no aerial mycelium. In an attempt to determine how many species were represented by these isolates, twelve isolates were co-inoculated in pairs in all possible combinations on plates of 3 % malt agar. After incubation at 22° C. the reaction, if any, at the junction between the two colonies was noted. It was assumed that no reaction was an indication of identity or close relationship. This assumption appeared to be justified by the absence of any reaction when two colonies of the same isolate were grown together. The reaction consisted of the formation of a line of densely intertwined hyphae in the agar, together with a heaping up of the aerial mycelium in the zone of contact. In some cases a brown pigment was formed in the agar at the point of contact between the two colonies. By recording the intensity of the reaction between pairs of isolates on a chequered diagram it was possible to group similar isolates together. In this way the twelve isolates were split up into three groups. Two of these groups were identical with two of the three groups mentioned previously and one was different. Combining these observations it appeared that four different species of Basidiomycetes were present.

The identity of the fungi

Such co-inoculation experiments, while revealing affinities between organisms, give no indication of the degree of relationship or difference. That is to say such physiological differences as are expressed by cultural behaviour may be differences between biotypes of the same species rather than between genera or species. Therefore attempts were made to induce the isolates to fruit so that the above groupings could be correlated with accepted taxonomic categories. Various methods were tried in the hope of producing fructifications, but without success. Subcultures of isolates B1, B8, B9 and B12 were kindly examined by Mr E. C. Badcock of the Forest Products Research Laboratory, Princes Risborough. He was able to induce isolates B1 and B12 to fruit on Norway Spruce sawdust medium (Badcock, 1941). The sporophores were identified by Miss E. M. Wakefield as *Corticum coronilla* v. Höhnel & Litsch. Mr Badcock also noted that isolate B9 was entirely different from isolates B1 and B12. Unlike them it would not grow on beech sawdust medium. Isolate B8, as mentioned above, was entirely different from the other fungi. The results of the co-inoculation tests were thus partly confirmed. B1 and B12 were there grouped together and were now shown independently to belong to the same species. The other isolates placed in this group may perhaps also be regarded as belonging to the species *Corticium coronilla*.

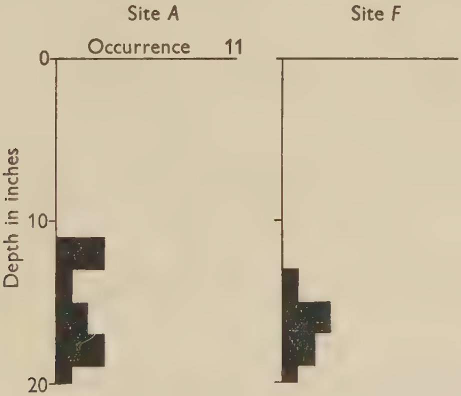


Fig. 3. Occurrence of Basidiomycetes in Wicken soils.

The distribution of the Basidiomycetes in the soil

The distribution of these fungi in the soil has several interesting features. They were isolated from sites *A* and *F* in approximately equal numbers. These sites were nearly 1 mile apart which suggests that these fungi have a wide horizontal distribution. Their vertical distribution (Fig. 3) is also of interest. At site *A* they are confined to a zone extending from 11 to 20 in. below the surface. They are most abundant at 11–13 in. and at 17–19 in. At site *F* their zone extends from 13 to 20 in. They are most abundant here at 15–17 in. These fungi would appear to be well-established members of the soil flora of this part of the profile. This is indicated by the regularity with which they were isolated over a period of 11 months. The average number of colonies recorded per sample is approximately the same as those of other fungi. At such depths at Wicken Fen the fungal flora is rather sparse.

The writer wishes to record his indebtedness to the late Prof. F. T. Brooks, F.R.S., who suggested and supervised this work. He also wishes to thank Dr J. H. Warcup for help and advice; and the following mycologists for help with the identification of some of the fungi: Mr E. C. Badcock, Dr K. B. Raper, Mr G. Smith, Dr M. B. Ellis, Dr S. J. Hughes and Dr C. G. Dobbs.

REFERENCES

Badcock, E. C. (1941). New methods for the cultivation of wood rotting fungi. *Trans. Brit. mycol. Soc.* **25**, 300–205.

Bisby, G. R., Timonin, M. I. & James, N. (1935). Fungi isolated from soil profiles in Manitoba. *Canad. J. Res.* C, **13**, 47–65.

Chesters, C. G. C. (1948). A contribution to the study of fungi in the soil. *Trans. Brit. mycol. Soc.* **30**, 100–117.

Chesters, C. G. C. (1949). Concerning fungi inhabiting soil. *Trans. Brit. mycol. Soc.* **32**, 194–216.

Corner, E. J. (1934). The fungi of Wicken Fen, Cambridgeshire. *Trans. Brit. mycol. Soc.* **19**, 280–287.

Godwin, H. (1929). The 'Sedge' and 'Litter' of Wicken Fen. *J. Ecol.* **17**, 148–160.

Godwin, H. (1931). Studies in the ecology of Wicken Fen. I. The ground water-level of the fen. *J. Ecol.* **19**, 449–473.

Godwin, H. & Barucha, F. R. (1932). Studies in the ecology of Wicken Fen. II. The fen water-table and its control of plant communities. *J. Ecol.* **20**, 157–191.

Godwin, H., Mobbs, R. H. & Barucha, F. R. (1932). Soil factors in Wicken Sedge Fen. *Nat. Hist. Wicken Fen*, pt. 51, 601–614.

Waksman, S. A. (1944). Three decades with soil fungi. *Soil. Sci.* **58**, 89–115.

Warcup, J. H. (1950). The soil plate method for isolation of fungi from soil. *Nature, Lond.*, **166**, 117.

Warcup, J. H. (1951*a*). The ecology of soil fungi. *Trans. Brit. mycol. Soc.* **34**, 376–399.

Warcup, J. H. (1951*b*). The effect of partial sterilization by steam or formalin on the fungus flora of an old forest nursery soil. *Trans. Brit. mycol. Soc.* **34**, 519–532.

Warcup, J. H. (1951*c*). Studies on the growth of Basidiomycetes in soil. *Ann. Bot., Lond.* (N.S.), **15**, 305–317.

Warcup, J. H. (1952). Effect of partial sterilization by steam or formalin on damping-off of Sitka spruce. *Trans. Brit. mycol. Soc.* **35**, 248–262.

(*Accepted for publication* 19 *September* 1952)

SOME SCOTTISH MOUNTAIN RUST FUNGI

By D. M. HENDERSON

Royal Botanic Garden, Edinburgh

Infection experiments were performed with *Melampsora epitea* from *Saxifraga hypnoides*, and with *Melampsora epitea* var. *reticulata* from *Saxifraga aizoides* to ascertain the uredo-, and the teleutospore hosts. The experiments showed that the rust on *S. hypnoides* infects *Salix herbacea* and some of its hybrids. The rust on *Saxifraga aizoides* infected only *Salix reticulata*.

The uredo paraphyses produced on *Salix reticulata* were larger than those for typical *Melampsora epitea* but fitted those of *M. epitea* var. *reticulata*. The uredo paraphyses on *Salix herbacea* and hybrids were typical of *Melampsora epitea*. *M. arctica* and *M. alpina* are considered indistinguishable from *M. epitea*. Observations show that *M. epitea* var. *reticulata* may perennate as mycelium in the *Saxifraga* host and the relation of this habit to dispersal and distribution in the field is mentioned.

Experimental studies of the rust fungi on arctic alpine plants have been rather neglected, no doubt on account of the difficulty of collecting and growing the host plants for infection experiments. During the last three years while collecting fungi on several of the higher Scottish mountains three species of *Saxifraga* bearing rust fungi were found. The relatively common microcyclic species, *Puccinia saxifragae* on *Saxifraga stellaris*, was collected on several occasions. In the summers of 1950 and 1951 spermogonia and caeomata of the *Melampsora* type were found on *Saxifraga aizoides* and *S. hypnoides*.

In recent years records of the occurrence of these species of *Melampsora* in Scotland have been summarized by Wilson (1934), whilst Jørstad (1940, 1951) has dealt with their distribution and biology in the northern hemisphere of the Old World. Both these authors give various species of *Salix* as the uredo- and teleutospore hosts of these saxifrage rusts, as do Sydow & Sydow (1915), Fischer (1904), Arthur (1925) and Grove (1913), but all these associations are based solely on field observations. The only successful experiments with species of *Melampsora* on saxifrage were carried out by Jacky (1899), who showed that the rust on *Saxifraga oppositifolia* produced its uredo- and teleutospore stages on *Salix herbacea*.

CAEOMATA ON *SAXIFRAGA HYPNOIDES*

There are few records of rust fungi on *S. hypnoides*. Dietel, in 1888, collected a rust on this host in the neighbourhood of Leipzig. Collections in Scotland on Ben Venue by Greville in 1888 and on Ben Lui by Wilson (1934) were assigned to *Caeoma saxifragarum* DC. by Wilson, as the uredo- and teleutospore stages were unknown. Larsen (1931) gave *Saxifraga hypnoides*, in addition to *S. caespitosa*, *S. aizoides* and *S. oppositifolia*, as a host for a fungus he called *Melampsora saxifragarum* (DC.) Schroeter in Iceland. Jørstad (1951) reports a rust on *Saxifraga hypnoides* as fairly common in Iceland. In June 1949 plants of *S. hypnoides* bearing rust sori were found at about 3000 ft. on Meall Garbh in the Ben Lawers range in Perthshire. The plants had just been released from their winter snow covering, but the spermogonia and caeomata were quite well developed. *Salix herbacea* was present in the neighbouring turf, but no rust sori could be found on the leaves of these plants. In 1950 a rust on the same host was collected at 2500 ft. in Caenlochan Glen at the head of Glen Isla in Perthshire. The area has been examined carefully, but the rust seems to be very scarce and has been found only in one narrow gulley—this in spite of the fact that the saxifrage is moderately abundant in the area. No willows could be found within a fifty yard radius of this station.

Infection experiments. As the fungus has been collected so seldom there have been few suggestions as to possible alternate hosts, but Jørstad suggests that *S. herbacea* serves this purpose in Iceland.

In 1951 infection experiments were carried out. Caeomata were collected from Glen Isla in July and the aecidiospores used to inoculate the following willows: *S. reticulata* L., *S. repens* L., *S. herbacea* L., *S. sadleri* (=*herbacea* × *lanata*), *S. repens* × *herbacea*, *S. lapponum* L., *S. lanata* L., *S. arbuscula* × *repens*, × *gilloti* A. G. Camus (=*lapponum* × *phylicifolia*), *S. myrsinites* × *phylicifolia*.

Two plants of each species were inoculated and were kept under bell jars for twenty-four hours. In order to prevent chance contamination the plants were grown in isolation.

The inoculations were carried out on 16 July. On 17 August two plants each of *Salix sadleri* and *S. herbacea* × *myrsinites* and one of *S. herbacea* bore uredo sori. As the species common to all these results, *S. herbacea*, was a likely host being common on the hills, and as the plants of *S. herbacea* originally inoculated were in poor growth, tests of the susceptibility of this willow were repeated. Uredospores were transferred from *S. sadleri* infected in the previous tests, to fresh plants of *S. herbacea* in August. Uredosori developed within three weeks and teleutospores had formed by autumn.

CAEOMATA ON *SAXIFRAGA AIZOIDES*

The caeomata on this host are quite common on certain of the higher Scottish hills, but it has been noticed that unless *Salix reticulata* is present in the area, although not necessarily in the vicinity of the saxifrage, the latter is not rusted. This observation is in agreement with the generally accepted association of the caeomata on *Saxifraga aizoides* and the uredo- and teleutospore stages on *Salix reticulata*. Hitherto, however, the association has not been proved experimentally, and has rested solely upon field observations. Juel (1911) experimented with this rust and obtained infection on *S. glauca* but apparently this may have been due to chance contamination. Lindfors (1913) inoculated *S. herbacea* and *S. lapponum* without success.

The same species of willow were used as in the previous experiment and the same procedure was followed. On 5 June the willows were inoculated with aecidiospores from *Saxifraga aizoides* collected on Creag an Lochan in the Breadalbane range. After fourteen days both plants of *Salix reticulata* bore uredosori and none of the other plants showed any sign of infection. The plants of *S. lanata* and *S. herbacea* used in this experiment were re-inoculated with freshly collected aecidiospores, but again they were not infected. Teleutospores developed freely on *S. reticulata* by autumn.

This experiment thus confirms the field observations of Jørstad and others that *S. reticulata* is an alternate host for the rust on *Saxifraga aizoides*. Jørstad (1951) records *Melampsora epitea* var. *reticulata* on various species of *Salix* from Iceland. These are probably specialized races, for the Scottish rust did not infect any species other than *S. reticulata*. The latter species does not occur in Iceland.

THE SYSTEMATIC POSITION OF THE SAXIFRAGE RUSTS

Rust on Saxifraga hypnoides

Spermogonia amphigenous, scattered, rather flat, approximately 200 μ in diameter, and 100 μ high. *Aecidia*, amphigenous and on the stem, scattered, surrounded by the ruptured epidermis, without any distinct marginal paraphyses, orange, 300–700 μ in diameter. *Spores* subglobose, 17–21 × 14–20 μ. *Spore wall* densely and minutely verruculose, 0·5–2 μ thick. On *S. hypnoides*.

Uredosori, hypophyllous, scattered, 0·5 mm. in diameter; *spores* 20·0 × 15·4 μ; *spore wall*, verruculose, 1·8 μ thick; *paraphyses* numerous, capitate, hyaline, up to 70 μ in length; heads of inner paraphyses 15·0–19·6 μ in diameter, wall 2·0–4·6 μ thick. *Teleutosori* amphigenous, subepidermal, reddish brown and waxy in appearance; *spores* brown, prismatic, not thickened at the apex, 23–46 × 6–16 μ. On *S. herbacea* (by inoculation).

The measurements of the uredo- and teleutospore stages on *S. sadleri* and *S. herbacea* × *myrsinites* are essentially similar to the above.

Two species of *Melampsora*, *M. alpina* Juel and *M. arctica* Rostrup, are given as occurring in *Salix herbacea* by Sydow & Sydow (1915) who, however, remark that there is little to distinguish them. Arthur agreed with this view and cites *Melampsora alpina* as a synonym of *M. arctica*.

The caeoma on *Saxifraga hypnoides* agrees with the description by Sydow of the caeoma of *Melampsora arctica* on *Saxifraga oppositifolia*. Lindfors (1913) examined the caeomata of several species of *Melampsora* and described elongate peripheral cells, swollen at the apex to the diameter of the spores in the caeoma of *M. cernuae*. Wilson (1919) described paraphyses in the caeomata of *M. arctica* on *Saxifraga oppositifolia*, but there is no reference to these structures in any other account of this fungus. There are no such cells in the caeomata on *S. hypnoides*.

The uredo- and teleutospore stages agree with those described for *Melampsora arctica*. Some, at least, of the collections of *Salix herbacea* made in this country and hitherto presumed to be stages of the fungus alternating with *Saxifraga oppositifolia* may thus equally well belong to the *S. hypnoides* rust. Whether the rusts on *Saxifraga oppositifolia* and *S. hypnoides* are specialized to their hosts is not known.

In an attempt to classify this group by morphology rather than host relationships Jørstad has suggested that all the *Salix* Melampsoras possessing subepidermal teleutosori with the spores not thickened at the apex and roundish to oval uredospores with uniformly echinulate walls, should be placed in the single species *Melampsora epitea* (Kunz & Schm.) Thuem. This name would include a large number of 'species' alternating between *Salix* and such taxonomically diverse hosts as *Larix*, *Saxifraga* and *Ribes*. The best course seems to be to accept the comprehensive specific epithet *Melampsora epitea*, and to consider the purely biological groups specialized to various aecidial hosts as specialized races, rather as Mains (1932) suggested for *Puccinia rubigo-vera*. The alternative to this course is the needless multiplication of rust species distinguishable only by their hosts.

Rust on Saxifraga aizoides

The caeoma on *S. aizoides* and the uredo- and teleutospore stages on *Salix reticulata* agree with those described by Jørstad for *Melampsora epitea* var. *reticulata* (Blytt) Jørstad. Jørstad (1940) distinguishes this variety as follows: in var. *reticulatae* the diameter of the heads of the inner paraphyses measure up to 25–35 μ whilst in typical *M. epitea* they do not usually exceed 21–22 μ. These characters hold good for the Scottish material, in which the heads measured 18–30 μ (21 μ).

THE PERENNATION OF *MELAMPSORA EPITEA* VAR. *RETICULATA*

The general problems of the survival and dispersal of rust fungi under arctic alpine conditions have been discussed by Lind (1927), Jørstad (1940) and Wilson (1948). These authors cite various modifications of the life cycle of these fungi to compensate for the disadvantages of the heteroecious habit expecially under conditions of short summers. The saxifrage rusts of the *M. epitea* type are suitable for investigations of these phenomena for they are by far the commonest heteroecious species in the Arctic (Arthur, 1928). Lind (1927), discussing the question of the wide distribution of some arctic species, suggested that they may be dispersed as perennial mycelium in fragments of the saxifrage host, blown over the ice and taking root in new localities. Lind mentions that the mycelium is perennial in *Saxifraga aizoides* as does Jørstad, but neither produce any evidence to support their statements. However, field observations in this country would support this view. It seems a general rule that *S. aizoides* may be infected a considerable distance from any plants of *Salix reticulata*, but not in areas where this willow is completely absent, which suggests that the aecidial phase may perennate independent of the dicaryon host. The distribution of aecidia on plants also suggests the existence of perennial mycelium. Isolated aecidial sori are not found. A group of completely infected stems is usually found, often arising within a mat of healthy stems. Furthermore, careful examination shows that all the stems of an infected group are connected. This distribution of infected stems in the midst of healthy ones does not suggest infection from outside during the current season especially when it is considered that the fructifications may be well developed only a few weeks after the snow has melted from the plants. All the field evidence points to perennation by mycelium in the saxifrage. To confirm this method of overwintering, infected plants were kept in isolation during the winter of 1951–2 in Edinburgh. By 20 March the spermogonia had developed in the young leaves, and examination showed that rust mycelium was present in the central pith of the current and previous year's growth. This confirms the general field observations of other authors. The explanation of the distribution of infected saxifrages is of some interest. As already noted, it seems a fairly general rule that *Saxifraga aizoides* may be infected a considerable distance from any plants of *Salix reticulata*, but not in areas where this willow is completely absent. If it be assumed that *S. reticulata* is the only dicaryon host in Britain then the saxifrage can only become infected in the neighbourhood of this willow, so that the distribution of the fungus will follow roughly that of the willow. However, once infected, portions of the saxifrage, and even whole plants, may be swept down mountain streams in spring floods and take root at lower levels where the fungus will exist solely by reason of the perennial mycelium in the saxifrage. Thus whilst the lower altitudinal limit of *S. reticulata* in central Scotland is approximately 2000 ft., infected plants of *Saxifraga aizoides* might be found at very much lower elevations. In 1951 caeomata on *S. aizoides* were collected at 800 ft. on Ben Lui. This explanation is somewhat similar to that given by Lind, except that whereas he postulates dispersal by wind-blown fragments over ice and snow, carriage by spring floods seems the most probable agent in this country.

My thanks are due to Dr Malcolm Wilson for first drawing my attention to these mountain fungi and for stimulating encouragement and advice.

REFERENCES

Arthur, J. C. (1928). Notes on arctic Uredinales. *Mycologia*, **21**, 41–43.

Dietel, P. (1890). Verzeichnis der in der Umgebung von Leipzig beobachteten Uredineen. *S.B. naturf. Ges. Lpz.* **37**, nos. 15 and 16.

Fischer, E. (1904). Die Uredineen der Schweiz. *Beitr. Kryptogamenfl. Schweiz*, **2**, 2.

Grove, W. B. (1913). *British Rust Fungi*. Cambridge.

Jacky, E. (1899). Untersuchungen über einige Schweizerische Rostpilze. *Ber. schweiz. bot. Ges.* **9**, 49.

Jørstad, I. (1940). Uredinales of Northern Norway. *Skr. norske VidenskAkad.* pt. 6.

Jørstad, I. (1951). The Uredinales of Iceland. *Skr. norske VidenskAkad.* pt. 2.

Juel, H. O. (1911). Meddelande vid Botaniska Sektionens of Naturvetenskapliga studentsallskupet sommarkomst den 30 Nov. 1910. *Svensk bot. Tidskr.* **5**, 232–233.

Larsen, P. (1931). The fungi of Iceland. *Botany of Iceland*, **2**, 452–607.

Lind, J. (1927). The geographical distribution of some arctic Micromycetes. *K. danske vidensk. Selsk.* **6**, 5.

Lindfors, Th. (1913). Aufzeichnungen über parasitische Pilze in Lule Lappmark. *Svensk bot. Tidskr.* **7**, 39–57.

Mains, E. B. (1932). Host specialization in the leaf rust of grasses, *Puccinia rubigo-vera*. *Pap. Mich. Acad. Sci.* **17**, 289–394.

Sydow, P. & Sydow, H. (1915). *Monographia Uredinearum*, **3**, Lipsiae.

Wilson, M. (1919). Some British rust Fungi. *J. Bot., Lond.*, **57**, 161–163.

Wilson, M. (1934). The distribution of the Uredineae in Scotland. *Trans. bot. Soc. Edinb.* **31**, 345–449.

Wilson, M. (1948). The occurrence in the Uredinales of species with unstable life history. *Proc. roy. Soc. Edinb.* B, **63**, 177–187.

(*Accepted for publication* 25 *September* 1952)

MICROMYCOPSIS OEDOGONII SP.NOV.

By MARJORIE ROBERTS

Birkbeck College, University of London

(With Plate 17 and 12 Text-figures)

Micromycopsis oedogonii sp.nov. parasitizing *Oedogonium* sp. is described and illustrated, and compared with previously recorded species of *Micromycopsis*, all of which attack members of Conjugales.

Micromycopsis oedogonii sp.nov.

Thallus in Oedogonii cellulis invenitur tumidis et in speciem oogonii globatis. Prosorus plerumque globosus 14–30 μ dia., nonnunquam autem praelongus (cum in adulta cellula insedit), tunica glabra, hyalina. Sporangiosorus epibioticus, plerumque sessilis, globosus, in 7–15 sporangia pyramidata divisus. Sori tunica brunea, aculeis minutis obtusis ornata. Zoospora subglobosa 2 μ diam., guttula oleosa unica, unico postico flagello munita.

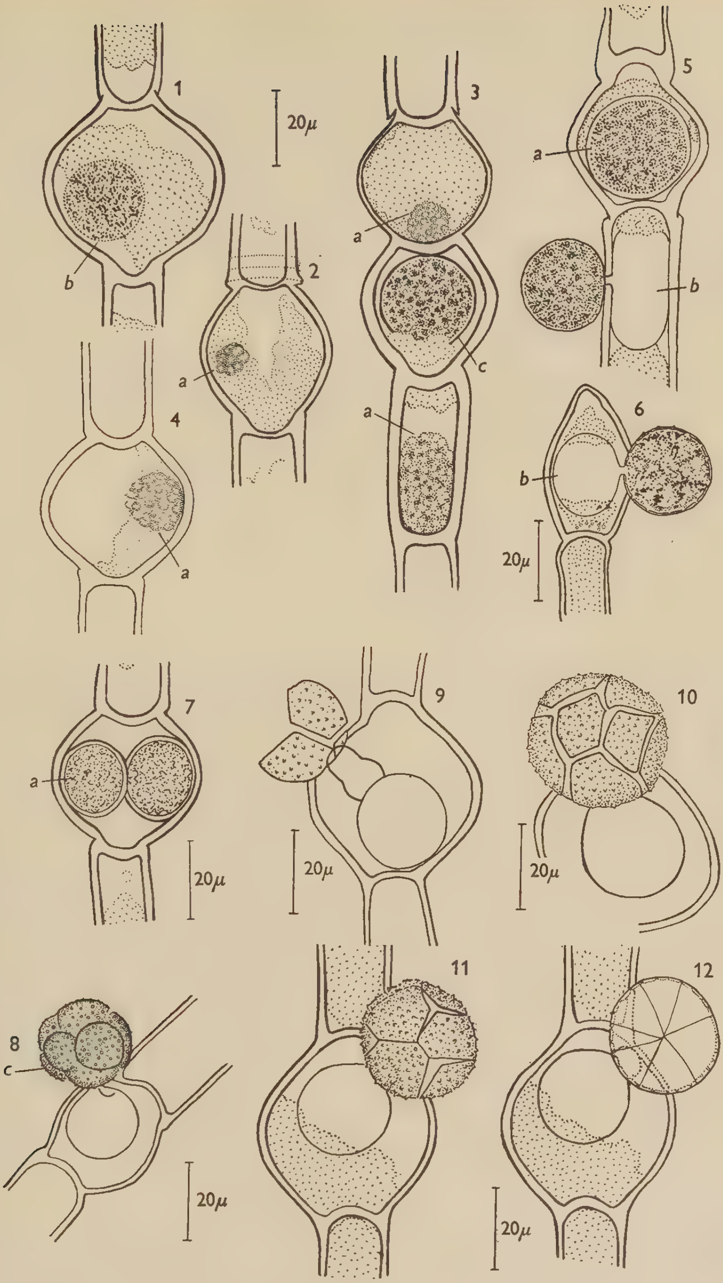
Hab. in *Oedogonio*, Bolder Mere, Wisley, Surrey, Angliae.

Type material deposited at Commonwealth Mycological Institute, Kew. (Herb. I.M.I. 50464.)

During the autumn of 1950 and the summer and autumn of 1951 *Micromycopsis oedogonii* was found in considerable abundance in collections of *Oedogonium* sp. from Bolder Mere, Wisley, Surrey, England. It seemed to be strictly limited to a single host species. This grew intimately mixed with other members of Oedogoniales (two species of *Bulbochaete* and two of *Oedogonium*, including *O. undulatum*) but these were never attacked, nor were species of *Zygnema* and *Mougeotia* which were sometimes in the collections. As winter approached the host became scarcer and the rise of the water level made collection difficult. The fate of the parasite during late winter and until its reappearance in the summer is unknown.

The fungus normally attacks the upper daughter cell formed by the peculiar cell division in *Oedogonium*. This cell, which initially has a very thin wall, is caused to swell until it resembles an oogonium (Text-fig. 2). Thalli of the parasite sometimes occur in unmodified cells, suggesting that these have been attacked at a stage when the walls have become relatively rigid. The thalli in such cells are elongated having obviously been forced into that shape by the narrow form of the host cell (Text-fig. 3). Thus cells containing elongated thalli are of normal form but always have thick walls, whilst in the hypertrophied host cells the wall is often quite thin, especially if the parasite is at an early stage of development (Text-fig. 3).

Before the parasite is seen, its presence can be inferred by the swelling of the host cell and by a change in the optical density of the chloroplast. When it first becomes visible the thallus is naked, somewhat irregular and appears to consist almost entirely of oil drops (Text-fig. 2). It is frequently



Text-figs. 1–12. *Micromycopsis oedogonii*

Text-figs. 1–4. Early stages. *a*, very young thalli at 'irregular' stage; *b*, thallus rounded off just before the formation of the prosorus wall; *c*, young prosorus. Host chloroplast shown by coarser and more regular stippling.

Text-figs. 5–8. Later stages. *a*, mature prosorus; *b*, empty prosorus with young sorus; *c*, sorus with mature sporangia.

Text-figs. 9–12. Text-fig. 9, empty prosorus and remains of sorus wall. Text-fig. 10, empty prosorus and sorus showing polygonal plates of sorus wall. Text-figs. 11 and 12, one specimen with sorus in surface view and optical section.

obscured by the remains of the chloroplast which at this stage is still green. The young thallus enlarges, becomes spherical and invested by a smooth, colourless wall whilst at the same time the oil drops cease to dominate the structure of the protoplasm which becomes granular and greyish. The parasite is now in the prosorous stage (Text-fig. 5), and the host chloroplast has disintegrated to a greater or lesser extent, having lost most of its colour. The mature prosorus contains protoplasm of a greyish brown colour, is surrounded by a smooth colourless cell wall and is normally spherical. The size distribution of 60 spherical prosori encountered at random is shown in Table 1.

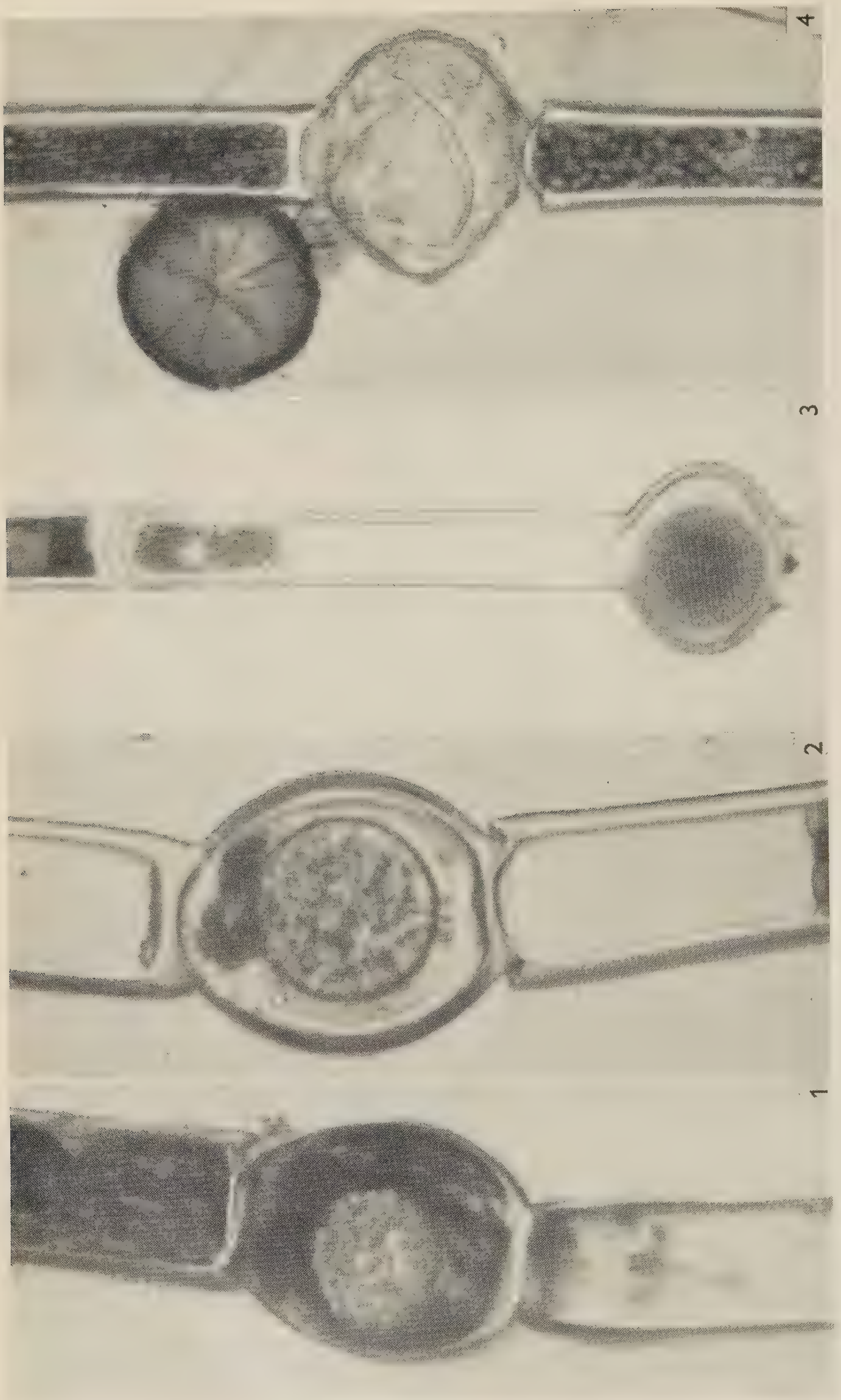
Table 1. *Prosorus size of* Micromycopsis oedogonii

Size range in μ	Number
> 14–16	4
> 16–18	10
> 18–20	10
> 20–22	16
> 22–24	7
> 24–26	8
> 26–28	4
> 28–30	1

Sometimes a double infection occurs and when this happens the two thalli may be at the same (Text-fig. 7) or different stages of development, within the same cell.

Early stages in the germination of the prosorus have not been observed, which is remarkable as so much living material has been examined. It appears, however, that an exit tube pierces the host wall and swells outside to form a spherical body which is sessile on the host (Text-fig. 5). Exit tubes vary in length from 2 to 13 μ. The external swelling becomes divided into 7–15 sporangia to form the sporangiosorus. Each sporangium appears triangular in optical section of the sorus (Text-fig. 12), but is really pyramidal with a curved external wall polygonal in surface view (Text-fig. 11). Thus the wall of the sporangiosorus appears to consist of a number of polygonal plates which are somewhat thickened, brownish and covered with short blunt spines. The internal walls of the sorus, separating the sporangia are thin and smooth. At maturity the sporangia swell slightly causing the plates of the sorus wall to separate to some extent (Text-fig. 8). At this stage the sporangiosorus looks rather like a blackberry. Only in a few examples were fully delimited zoospores seen in the sporangia, but each seemed to contain about 50.

The liberation of zoospores was seen only once and it was not clear if the escape was through a pore in the thin part of the sporangium wall, as reported for other species. The zoospores were sub-spherical, about 2 μ in diameter, each with a conspicuous oil drop and a single posterior flagellum. They were zoids of the characteristic chytridian type. In this species there appears to be only one kind of zoospore in the life cycle, not two as reported by Canter (1949) in *Micromycopsis fischeri* and as suggested by Scherffel (1926) in *M. cristata*. Infection of host filaments with zoospores was not observed.


4
3
2
1

Since *M. oedogonii* can apparently be found only for a few months each year, there is the problem of its survival during the rest of the year. No specialized resting spores were seen, but it is likely that certain prosori function as such. This is in conformity with what is known of other aquatic members of Synchytriaceae.

Micromycopsis oedogonii is distinguished sharply from other known species of aquatic Synchytriaceae in parasitizing *Oedogonium* sp. Previous records (Table 2) name desmids or filamentous Conjugales as hosts. Morphologically *Micromycopsis oedogonii* resembles *M. intermedia* most closely, but differs in having a larger number of sporangia in the sorus, and in having a prosorus wall that is colourless and never spiny. It differs from *M. fischeri* in having only one type of zoospore, the typical chytrid zoid liberated directly from the sporangia of the sorus, and from *M. cristata* in its pyramidal sporangia.

Table 2. *List of aquatic Synchytriaceae with recorded hosts based on information in Sparrow* (1943) *and Canter* (1949)

Species	Host
Endodesmidium formosum	*Netrium oblongum, Cylindrocystis* spp.
Micromyces zygogonii	*Zygogonium* sp., *Spirogyra quadrata, Mesocarpus scalaris, Netrium* sp., *Mougeotia* spp.
M. petersenii	*Mougeotia* sp.
M. longispinus	*Spirogyra* sp.
M. laevis	*Mougeotia* sp.
Micromycopsis cristata	*Hyalotheca* spp., *Spirogyra* sp., *Zygogonium* sp.
M. fischeri	*Zygogonium* sp., *Tetmemorus brebissonii*
M. zygnemicola	*Zygnema stellina*
M. intermedia	*Zygnema* spp.
M. mirabilis	*Closterium* spp.

My thanks are due to Prof. D. Jones for help with the Latin diagnosis and especially to Professor C. T. Ingold under whose direction this investigation was carried out.

REFERENCES

Canter, H. (1949). Studies on British Chytrids. VI. Aquatic Synchytriaceae. *Trans. Brit. mycol. Soc.* **32**, 69–94.

Scherffel, A. (1926). Einiges über neue oder ungenügend bekannte Chytridineen. *Arch. Protistenk.* **54**, 167–260.

Sparrow, F. K. (1943). *Aquatic Phycomycetes.* Ann Arbor, U.S.A.

EXPLANATION OF PLATE 17

Micromycopsis oedogonii

Fig. 1. Naked thallus inside hypertrophied *Oedogonium* cell. Chloroplast still large and green. × 860.

Fig. 2. Mature prosorus. Chloroplast of swollen host cell largely disintegrated. × 970.

Fig. 3. Two infected cells of an *Oedogonium* filament. Upper cell not hypertrophied with an elongated prosorus in its upper end. Lower cell hypertrophied with an oospore-like prosorus. × 720.

Fig. 4. Swollen host cell containing empty prosorus partially collapsed. Outside is the sporangiosorus seen in optical section showing the individual triangular sporangia. × 690.

(*Accepted for publication 2 October* 1952)

CROWN ROT, A SEEDLING DISEASE OF GROUNDNUTS CAUSED BY *ASPERGILLUS NIGER*

II. AN ANOMALOUS EFFECT OF ORGANO-MERCURIAL SEED DRESSINGS

By I. A. S. GIBSON

*Botany School, University of Cambridge**

Field experiments carried out in East Africa have shown that seed dressings with Thiram (tetramethylthiuram disulphide) preparations may reduce the incidence of crown rot in groundnut (*Arachis hypogea* L.) seedlings by at least 50%. Seed treatment with organo-mercurials, on the other hand, has generally produced a substantial increase in the incidence of crown rot.

Experiments designed to elucidate this effect of organo-mercurial seed dressings have indicated that it cannot be ascribed to any phytotoxic action. The effect can probably be attributed to the selectively toxic action of organo-mercurials upon the fungus flora of the soil in the vicinity of the treated seed.

This selective toxicity has been shown to operate to the advantage of *Aspergillus niger*, certain strains of which have proved to be much more tolerant of organo-mercurials than other fungi of Urambo soil. Tolerant species of *Penicillium* have occasionally been encountered. Mercury tolerant strains of *Aspergillus niger* have been isolated from Urambo soil; they are distinguished from mercury sensitive strains both by a greater and more rapid development of acidity in the culture medium, and by a greater virulence in the production of crown rot when inoculated to groundnut seedlings.

Introduction

During 1948–51, while on the staff of the Overseas Food Corporation at Urambo, Tanganyika Territory, I obtained unexpected results in a series of field trials designed to test the efficiency of various fungicidal seed dressings for the control of seed diseases of groundnut (*Arachis hypogea* L.). These seedling diseases comprise: (*a*) seed rotting and pre-emergence killing of seedlings caused mainly by *Rhizopus* spp.; and (*b*) crown rot, the chief cause of post-emergence killing and also causing a small proportion of pre-emergence killing, due to infection of the seedling hypocotyl by *Aspergillus niger* van Tiegh. (Gibson & Clinton, 1953; Gibson, 1953). Seed rotting and pre-emergence killing can be greatly reduced, though not entirely eliminated, by dressing the seed either with an organo-mercurial or with various other fungicides such as Thiram (tetramethylthiuram disulphide) (Gibson & Clinton, 1953). In general, the organo-mercurial preparations are more effective in reducing such pre-emergence losses than any other type of seed dressing. The effects of seed dressings on the incidence of crown rot, obtained in trials carried out over a period of 3 years at Urambo and Kongwa, are presented below (Tables 1 and 2).

---

* Formerly of the Overseas Food Corporation; now in the Forestry Department, Nairobi, Kenya.

Whereas the Thiram preparations consistently reduced the incidence of crown rot, the organo-mercurial dressings generally increased it, sometimes to a marked degree. The fact that this unwelcome effect of the organo-mercurial treatment is not entirely consistent in its intensity and incidence (it is scarcely apparent, for instance, in the trial of organo-mercurial A in 1948–9, Table 1), is readily explicable on the hypothesis that this class of fungicide influences incidence of crown rot in several distinct ways, and that its effect on disease incidence varies according to the relative preponderance of one or other of these different effects (see below).

Table 1. *The effects of organo-mercurial and other seed dressings on the incidence of crown rot at Kongwa and Urambo*

Preparation	Rate of application (oz./cwt.)	Crown rot incidence (per 1000 plants)				
		Urambo			Kongwa	
		1948–9	1949–50	1950–1	1949–50	1950–1
Nil	—	29·7	24·1	19·9	16·2	8·9
Organo-mercurial A	2·0	34·8	62·7	37·5	20·7	41·7
	4·0	24·4	47·5	53·9	18·2	51·4
	6·0	—	—	44·9	—	43·7
	8·0	17·9	46·7	—	17·6	—
Thiram C	1·0	—	—	9·4	—	—
	2·0	—	—	8·0	—	—
	4·0	—	—	9·7	—	—
Mercury preparation A	3·2	—	—	—	—	22·7
	6·4	—	—	—	—	14·2
	9·6	—	—	—	—	12·2

Table 2. *The effects of organo-mercurial and other seed dressings on the incidence of crown rot at Kongwa and Urambo*, 1949–50

Preparation	Rate of application (oz./cwt.)	Crown rot incidence (per 1000 plants)	
		Urambo	Kongwa
Nil	—	15·3	32·2
Organo-mercurial A	2·0	24·7	46·6
	6·0	27·3	—
Organo-mercurial B	2·0	51·4	33·2
	6·0	21·8	—
Organo-mercurial C	3·0	31·8	33·2
	8·0	22·8	—
Organo-mercurial D	100*	23·5	25·2
	200*	10·8	—
Thiram A	1·5	7·5	18·4
	3·0	7·5	—
Thiram B	1·5	6·3	5·9
	3·0	8·5	—

* ml./cwt.

First, however, it is necessary to consider the possibility that this apparent increased incidence of crown rot in seedlings from organo-mercurial dressed seed is not a true increase in incidence of *total* infection by *Aspergillus niger*. Thus the seed treatment might reduce the 'inoculum potential' of *A. niger* on or around the groundnut seed in such a way that a reduction in the aggressiveness of infection results in a lower incidence of pre-emergence mortality, and that the higher post-emergence mortality represents a 'deferred instalment' of pre-emergence mortality. The seed treatment would merely defer the killing of the seedling by *A. niger*.

Without further evidence from field experiments specifically designed to test this possibility (which had not been anticipated when the original series of field experiments was planned), it is impossible to disprove this hypothesis, which may indeed account for part of the observed effect. Nevertheless, if this were the whole explanation, it would be difficult to account for the complete absence of any such effect with Thiram and other non-mercurial seed dressings, which substantially reduce both the total pre-emergence killing and the post-emergence crown rot.

It therefore seemed worth while to consider more specific ways in which organo-mercurial seed dressings might increase the incidence of post-emergence crown rot. There are two ways in which seed treatment might produce such an effect, and both these are open to experimental testing.

(1) The organo-mercurial may produce a partial sterilization effect on the seed-surface flora and on the microflora of the surrounding soil, due to the well-known fact that *A. niger* is more resistant to the action of fungicides and toxins than are many other soil fungi. The reduction in competition in this way could increase the 'inoculum potential' of *A. niger*.

(2) A phytotoxic effect of the organo-mercurial may reduce seedling resistance to infection by *A. niger*.

The occurrence of either or both of these effects does not, however, exclude the possibility that the organo-mercurial also exerts an opposing, or beneficial, effect on control of crown rot by the killing of *A. niger* inoculum in immediate proximity to the seed. From the available evidence (Morwood, 1945, 1946) it appears that under some conditions control of crown rot may be achieved by the use of organo-mercurial seed dressings. There is evidence (Gibson, 1953) that inoculum for hypocotyl infections may be provided by: (*a*) mycelium and spores of *A. niger* carried on the seed; and (*b*) organic debris colonized by the fungus. Infected or contaminated seed can give rise to infected cotyledons, which in turn infect the hypocotyl. It seems reasonable to assume that the fungicidal and fungistatic effects of the organo-mercurial upon *A. niger* must increase with proximity to the treated seed, whereas the stimulating effect of the treatment upon infection by *A. niger* is more likely to operate upon inoculum at some distance from the treated seed. The experiments to be described below were designed to test these various possibilities.

THE EFFECT OF ORGANO-MERCURIAL PREPARATIONS ON INOCULUM OF *ASPERGILLUS NIGER* CARRIED ON THE SEED

Different lots of groundnut seed were inoculated with spore suspensions of *A. niger* at the following concentrations: $1{\cdot}5 \times 10^4$, $1{\cdot}5 \times 10^6$, and $1{\cdot}5 \times 10^7$ spores/ml. A fourth lot was left uninoculated as a control. After inoculation, one half of each lot of seed was treated with organo-mercurial seed dressing A, at a rate of 2 oz./cwt. seed, and the other half left untreated. Seeds were planted in sand in boxes maintained in the glasshouse: temperatures during the experiment were comparable to those obtaining during the growing season in East Africa (max. 95° F., min. 70° F.).

Seedlings were counted for estimation of pre-emergence losses and scored for incidence of crown rot, which developed from infected cotyledons and caused the death of some plants (Table 3).

Table 3. *The effects of seed dressing on pre-emergence killing and post-emergence crown rot due to* Aspergillus niger *as a seed contaminant*

	No seed dressing		Seed dressing with organo-mercurial A at 2 oz./cwt.	
Inoculum concn., spores/ml.	Emergence (% seed planted)	Crown rot (% emerged seedlings)	Emergence (% seed planted)	Crown rot (% emerged seedlings)
$1{\cdot}5 \times 10^7$	53	77	51	59
$1{\cdot}5 \times 10^6$	58	63	53	55
$1{\cdot}5 \times 10^4$	49	71	67	43
Nil	96	20	96	7

From these results it appears that the organo-mercurial dressing has reduced pre-emergence killing by *A. niger* only at the lowest inoculum level. Incidence of post-emergence crown rot has been reduced by seed dressing at all inoculum levels, and most markedly at the lowest levels. The results thus suggest that where inoculum of *A. niger* is present only in contact with the seed surface, the beneficial fungicidal action of the seed dressing outweighs any disadvantageous effect in assisting infection by any surviving spores or mycelium of *A. niger*. To explain the field results (Tables 1 and 2), therefore, it seems necessary to postulate the occurrence of effects on inoculum at some distance from the seed.

THE EFFECT OF ORGANO-MERCURIAL PREPARATIONS ON *ASPERGILLUS NIGER* IN AFRICAN (URAMBO) SOIL

The following series of experiments were carried out to test the hypothesis, that organo-mercurial preparations may exert a selective or partially sterilizing action on the soil microflora, in such a way as to encourage the multiplication and activity of *A. niger*. In the first experiments a modified form of the Heatley cup plate technique was employed (Heatley, 1944), in which the cup containing the test substance was replaced by an agar plug incorporating a thick suspension of the commercial fungicide in the form employed in field practice. This plug, cut by a cork-borer (8·5 mm.) from a plate of 50 : 50 fungicide and 2 % agar mixture, was placed in a corresponding hole cut in an agar suspension of Urambo soil. (A similar technique has been employed by Palmiter & Keitt (1937) to test the toxicity of copper-lime-arsenite mixtures.) This soil suspension was prepared by Warcup's method (Warcup, 1950). After incubation of these soil plates for 48 hr. at 25° C., a zone free of fungus development around the fungicide plug became apparent. The size of this zone varied with the type of fungicide employed, but was constant for a given fungicide, providing sufficient active material was incorporated in the plug to establish a saturated solution in its immediate vicinity. Preliminary observations were made using this technique with the object of tracing differences in tolerance to fungicides between various common members of the Urambo soil fungus flora. It was expected that such a difference might be revealed by a zonation of fungal species at increasing distances from the plug. In such a test with organo-mercurial A and Thiram C, the inhibition zones established by the organo-mercurial were much larger than those with the Thiram preparation. No marked zonation of fungus types leading away from either fungicide was observed. However, after incubation of these plates for a further 48 hr., the small inhibition zone around the Thiram plugs was observed to be colonized by *Rhizopus* spp. *Aspergillus niger* was not seen as a colonist around these plugs. On the other hand, in plates with organo-mercurial plugs, the inhibition zones remained bare of fungal growth except where colonies of *A. niger* had arisen on the margin of the zone, when a vigorous growth of this fungus sometimes developed across the zone around the fungicide plug. Table 4 gives the results of counts for the frequency of *A. niger* colonies arising at: (1) the zonal margin; and (2) outside the inhibition zone of an organo-mercurial fungicide plug.

Table 4. *The frequency of colonies of* Aspergillus niger *in twelve test plates with Urambo soil and organo-mercurial A*

A. niger colonies as percentage of total fungus colonies			
At zone margin	Outside inhibition zone	Total no. of *A. niger* colonies at margin	Total no. growing into inhibition zone
22	23	20	3

In other experiments, the percentage of *A. niger* colonies growing into the inhibition zone has been greater (up to 50 %). From these observations it appeared that in the relatively wide zone around the organo-mercurial plug the concentration of the fungicidal agent was sufficient to prevent the germination of spores or mycelial fragments of any fungi present in Urambo soil, including those of *A. niger*. The concentration of fungicide was not sufficient, however, to prevent the growth into the bare zone of mycelium from certain already established colonies of *A. niger*, provided that such colonies had arisen sufficiently near the margin of the zone to gain access to it. It seems evident, therefore, that the growing mycelium of such colonies can tolerate a concentration of organo-mercurial that will inhibit germination of spores or mycelial fragments of the same species; this conclusion has been verified by the next experiments to be described. It might be thought that a similar explanation would account for the analogous growth of *Rhizopus* spp. into the narrower bare zones formed around the Thiram plugs, but further consideration suggests an alternative explanation. Thus it is equally possible that the mycelium of various soil fungi can advance across the narrow (and possibly less toxic) zones around the Thiram plugs, but that *Rhizopus* spp. are the most common colonists merely because the mycelium of these fungi grows very rapidly. They are typical 'saprophytic sugar fungi', which are the commonest first colonizers of sugar substrates owing to their high growth rate (Garrett, 1951). This explanation finds a close analogy in the observations of Walker (1941) in his study of fungal colonization of wheat straw. When the pieces of wheat straw were treated with calcium hypochlorite, which is a mild and non-selective surface sterilizing agent, *Fusarium culmorum* (a fungus of rapid and vigorous growth habit) was the commonest fungus growing out from the treated straws on to the agar plates, just as it was when the straws were merely washed with sterile water. But when the straws were given prolonged treatments with mercuric chloride or silver nitrate, species of *Penicillium* were virtually the only fungi surviving and able to colonize the agar from the treated straws.

The next series of experiments was designed to test the conclusion that *Aspergillus niger* is more tolerant to organo-mercurial A than are the majority of other soil fungi, and also to see whether growing mycelium of this fungus will tolerate concentrations of the fungicide that inhibit germination of mycelial fragments or conidia. For each series of experiments a dilution series of an aqueous extract of the commercial fungicide preparation was used, made up in 2 % dextrose Dox agar. In some experiments, Urambo soil was incorporated with the agar by Warcup's method; in others, the plates were inoculated either with conidia or with agar culture disks of *A. niger*. At the highest concentrations of organo-mercurial A, *A. niger* and a species of *Penicillium* alone survived in the Urambo soil plates. In the case of Thiram C, however, no modification of the proportions of the fungi appearing on any of the plates was found for any strength of fungicide up to 1/10 of saturation. When mycelial and conidial inocula of *Aspergillus niger* were compared under similar conditions, growth from conidia was completely stopped at 1/320 dilution of organo-mercurial A extract, whereas that from agar mycelium disks was inhibited only at a dilution of 1/40. No such difference was observed in the case of Thiram C, where growth from conidia and from mycelium were both reduced by 50 % at a dilution of 1/20.

VARIATION IN MERCURY TOLERANCE OF DIFFERENT STRAINS OF *ASPERGILLUS NIGER*

Isolations were made from those colonies of *A. niger* that were apparently tolerant or sensitive to organo-mercurial A. These were later tested for this character by suspending conidia from the different isolates in melted but cooled 2 % dextrose-Dox agar and inserting the standard fungicide-agar plug in the same way as for soil plates. After incubation of these plates for 48 hr. at 25° C., the relative sizes of the inhibition zones formed agreed with expectation according to the mode of origin of each isolate. Furthermore, the fungus colony from tolerant isolates continued to grow towards the fungicide plug, whereas the size of the inhibition zone of the sensitive isolates remained the same with further incubation (see Table 5).

Table 5. *Mean sizes of inhibition zones due to organo-mercurial A acting on tolerant and sensitive isolates of* Aspergillus niger

		Time after inoculation (days)			
Isolate and character		3	5	7	9
40, tolerant	Radius of zone (cm.)	2·4	2·0	1·7	1·4
6T, tolerant		2·6	2·0	1·7	1·4
7, sensitive		5·5	5·6	5·7	5·6
6S, sensitive		5·5	5·5	5·4	5·5

Following these tests, experiments were carried out to determine the relative strengths of organo-mercurial A and mercuric chloride required to inhibit the growth of mycelium and spores of tolerant and sensitive isolates of *A. niger*. For this purpose, dilution plates were prepared from an aqueous extract of the fungicide and from a solution of mercuric chloride respectively. Mycelial and conidial inocula were used as described previously, and the lowest concentration of fungicide at which growth just failed to occur was noted in each case.

The results showed that the concentration of fungicide required to inhibit growth from mycelial inoculum was always higher than that required to inhibit growth from sporal inoculum. The ratio between these two concentrations was 40 for tolerant strains with organo-mercurial A and 5 for the same strains with mercuric chloride; for sensitive strains the ratio was 5 with both fungicides. Both with organo-mercurial A and with mercuric chloride there were pronounced differences in the concentration at which spore-germination of tolerant and sensitive isolates was inhibited. With both reagents, spores from tolerant isolates withstood concentrations of fungicide 25 times stronger than those tolerated by spores of sensitive isolates. Further experiments have shown that isolates selected for tolerant or sensitive reactions to organo-mercurial A show the same characteristics with regard to other organo-mercurial preparations, and vice versa.

Tests carried out to determine the extent of acid production by these isolates have shown that tolerant isolates produce more titratable acid and a much lower pH value in liquid culture than do sensitive isolates (Table 6).

Table 6. *Relative acid production by tolerant and sensitive isolates of* Aspergillus niger *in liquid culture at* 25° C.

Isolate	Acid titre after 130 hr. incubation in 2 % dextrose Dox $(N \times 10^{-2})$	pH of medium after 36 hr. growth (initial pH of medium 5·4)	
		2 % dextrose Dox	0·2 % dextrose Dox
Tolerant			
1 T	3·7	<3·6	<3·6
2 T	3·8	<3·6	<3·6
3 T	3·4	<3·6	<3·6
4 T	2·0	<3·6	<3·6
5 T	4·3	—	—
6 T	4·4	—	—
7 T	—	4·1	<3·6
Sensitive			
1 S	1·2	4·8	>5·2
2 S	1·6	>5·2	>5·2
3 S	1·3	4·8	>5·2
4 S	1·4	—	—
5 S	2·9	>5·2	>5·2
6 S	—	>5·2	>5·2

The high degree of acidity resulting from the mycelial growth of tolerant isolates of *A. niger* may directly affect the activity of organo-mercurial compounds in solution. Similar effects have been recorded elsewhere (Albert, 1951), in which the degree of ionic dissociation may greatly change the toxicity of a given fungicide; such a change in degree of dissociation may be brought about by a change in pH of the medium.

The role of oxalic acid as an adjuvant to the invasion of ground-nut hypocotyls by *A. niger* in cases of crown rot has been discussed by Gibson (1953). Taken in conjunction with the results in Table 6, this suggests that isolates of *A. niger* that are tolerant to organo-mercurial compounds may also show a greater pathogenic activity in the production of crown rot. A series of experiments has been carried out to test this possibility, using a technique already described (Gibson, 1953), in which plants are grown in sand under artificial light at a constant temperature of 30° C. A summary of the results of these experiments is given in Table 7.

Table 7. *Pathogenicity of tolerant and sensitive isolates of* Aspergillus niger

	Isolates tolerant to organo-mercurial A			Isolates sensitive to organo-mercurial A		
Exp. no.	No.	Pathogenicity rating	Mean	No.	Pathogenicity rating	Mean
1	15	92	86	7	61	30
	29	81		28	27	
				34	3	
2	2 T	57	45	1 S	4	14
	5 T	54		3 S	21	
	6 T	23		6 S	16	
3	1 T	29	24	2 S	14	10
	3 T	41		4 S	7	
	4 T	13		5 S	10	
	7 T	11				
	8 T	2				
	4 A	24				
	1 B	8				
	3 B	41				
	5 B	46				

The results of these experiments confirm the conclusion that under circumstances leading to selection for tolerance of *A. niger* to organo-mercurial fungicides, there is also a tendency for the selection of strains showing increased pathogenicity.

EXPERIMENTS TO TEST THE EFFECT OF ORGANO-MERCURIAL COMPOUNDS ON THE SOIL FUNGUS FLORA IN A SOIL ENVIRONMENT

A final experiment was carried out to test the selective effects of organo-mercurials observed *in vitro* under soil conditions which approached those in nature as closely as possible. Urambo grey loam soil was brought to near-saturation with water after packing into glass tubes 8 in. long by $\frac{3}{4}$ in. diameter. The tubes were completely filled with soil, and were plugged at one end with a rubber bung and at the other with cotton-wool. Groundnut seeds, killed by exposure to dry heat at 90° C., were planted singly in the soil tubes at the end plugged with cotton-wool. Half of the tubes were planted with seed that had received no fungicide treatment after killing, and the other half received seed dusted with organo-mercurial A at 3 oz./cwt. The tubes were incubated at 25° C., and sampled at intervals of 23, 66, and 164 hr. after setting-up; samples were taken from soil in contact with the seed and at distances of 2 and 6 cm. from it. Duplicate tubes were taken at each time interval from those containing dusted and undusted seed; these were discarded after sampling. From each sample, four Warcup soil plates were prepared for estimation of the *A. niger* flora of the soil (Table 8).

Table 8. *Effect of seed dressing with organo-mercurial A on the* Aspergillus niger *flora of the surrounding soil*

Distance of samples from seed (cm.)	Undusted seed. Sampling times (hr.)			Dusted seed. Sampling times (hr.)		
	23	66	164	23	66	164
No. of *A. niger* colonies per plate expressed as percentage total fungus colonies						
0	34	35	11	48	38	17
2	18	18	46	40	25	42
6	47	22	47	42	21	48
Area of *A. niger* colonies expressed as percentage total fungus growth						
0	28	43	—	56	70	—
2	8	9	—	31	24	—
6	14	14	—	17	19	—

These results show that, as might have been anticipated, no effect of the organo-mercurial seed dressing on samples of soil taken at a distance of 6 cm. from the seed can be discerned. In soil samples taken at distances of 0 and 2 cm. from the seed, however, several effects of the seed treatment can perhaps be perceived, in spite of variability due to inadequate replication of sampling. In the first place, the numerical proportion of *A. niger* colonies to total fungus colonies (expressed in Table 8 as percentages) on the soil plates is increased by the seed treatment at the earlier sampling (23 and 66 hr.), but after 164 hr. the proportion is scarcely different from that in the soil samples from tubes with untreated seed. This suggests that the organo-mercurial diffusing into the soil around the treated seed has delayed the appearance of visible colonies of other fungi to a greater extent than that of *A. niger* colonies, thus sometimes resulting in a higher proportion of *A. niger* colonies at the two earlier samplings. The temporary advantage thus given to *A. niger* in the colonization of the soil plates is also reflected by an increase in the area of fungal colonization occupied by *A. niger*; this increase in proportional area of the plate covered by *A. niger* with soil samples from the organo-mercurial series is clearly shown in the lower half of Table 8, and supports the interpretation of the effect on percentage number of colonies.

These results thus support the general hypothesis that the partial soil sterilization produced by organo-mercurial seed treatment operates to the advantage of *A. niger* mycelium in the surrounding soil. This effect is analogous to that produced by sterilization of soil with carbon disulphide and formalin, in promoting dominance of the soil fungus flora by *Trichoderma viride*, as described by Bliss (1951) and Warcup (1951, 1952).

PHYTOTOXIC EFFECTS OF ORGANO-MERCURIAL A ON THE SEEDLING GROUNDNUT

Two series of experiments have been carried out in an attempt to demonstrate possible phytotoxic effects of organo-mercurial dressings on seedling groundnuts which might lead to a greater susceptibility of the host plant to attack by *Aspergillus niger*. In the first series, groundnut seeds were germinated on filter-paper soaked in a dilution series of aqueous extract of organo-mercurial A: phytotoxic effects were traced by periodical measurements of rootlet length. At the same time growth of *A. niger* was measured in cultures set up in agar containing the same dilution series of the fungicide: as in previous experiments of this nature, both mycelial and conidial forms of inoculum were employed. From these experiments it was observed that while phytotoxic effects only occurred at the highest concentrations of the fungicide, growth from conidial inoculum of *A. niger* was initiated only at much lower levels: the mycelial inoculum, on the other hand, was able to grow at concentrations of the fungicide which gave slight phytotoxic effects on the host plant. The second series of experiments was carried out in the glasshouse with groundnut seeds planted in sand; half of these seeds had received treatment with organo-mercurial A at a rate of 4 oz./cwt. of seed, while the other half was left untreated. For each of the two treatments, 288 seeds were planted. On emergence, when the plants were at the two- to four-leaf stage, all plants were inoculated with agar culture disks of *A. niger* on the hypocotyl. Counts of seedling emergence were made, and at the end of the experiments plants were scored for disease ratings. There was no significant difference between treatments with respect either to seedling emergence or to disease incidence. It is concluded from these observations that any slight phytotoxic effects that may accrue from seed treatment with organo-mercurial A under natural conditions would play very little part in the increase in incidence of crown rot.

The completion of this investigation was rendered possible through the generous provision of a research grant by the Overseas Food Corporation, to whom I am indebted. I also wish to express my appreciation of the help of Dr S. D. Garrett, who has supervised the work carried out in the United Kingdom. Acknowledgement is also due to Dr J. R. Booer and Mr R. V. Tipler, for their advice with regard to organo-mercurial compounds.

REFERENCES

ALBERT, A. (1951). *Selective Toxicity*. London: Methuen and Co.

BLISS, D. E. (1951). The destruction of *Armillaria mellea* in citrus soils. *Phytopathology*, **41**, 665–683.

GARRETT, S. D. (1951). Ecological groups of soil fungi: a survey of substrate relationships. *New Phytol.* **50**, 149–166.

GIBSON, I. A. S. (1953). Crown rot, a seedling disease of groundnuts caused by *Aspergillus niger*. *Trans. Brit. mycol. Soc.* **36**, 198–209.

GIBSON, I. A. S. & CLINTON, P. K. S. (1953). A study of pre-emergence seed-bed losses in groundnut cultivations of the Overseas Food Corporation in Tanganyika, East Africa. *Emp. J. exp. Agric.* (in the Press).

HEATLEY, G. (1944). A method for the assay of penicillin. *Biochem. J.* **38**, 61–66.

Morwood, R. B. (1945). Peanut diseases. *Qd agric. J.* **61**, 266–271.

Morwood, R. B. (1946). Peanut crown rot. *Qd agric. J.* **63**, 18–19.

Palmiter, D. H. & Keitt, G. W. (1937). The toxicity of copper-lime-arsenite mixtures to certain phytopathogenic fungi grown on malt agar plates. *J. agric. Res.* **55**, 439–452.

Walker, A. G. (1941). The colonization of buried wheat straw by soil fungi with special reference to *Fusarium culmorum*. *Ann. appl. Biol.* **38**, 333–350.

Warcup, J. H. (1950). The soil plate method of isolating fungi from the soil. *Nature, Lond.*, **166**, 117–118.

Warcup, J. H. (1951). Effect of partial sterilization by steam or formalin on the fungus flora of an old forest nursery soil. *Trans. Brit. mycol. Soc.* **34**, 519–532.

Warcup, J. H. (1952). Effect of partial sterilization by steam or formalin on damping-off of Sitka spruce. *Trans. Brit. mycol. Soc.* **35**, 248–262.

(*Accepted for publication* 1 *November* 1952)

SOME FUNGUS DISEASES OF *DIGITALIS LANATA*

By J. F. SPILSBURY

The Wellcome Research Laboratories, Beckenham, Kent

(With Plates 18, 19 and 20, and 5 Text-figures)

During the summer of 1952, three diseases caused by fungi have been observed on *Digitalis lanata* Ehrh. resulting from infection by *Colletotrichum fuscum* Laubert, *Septoria digitalis* Pass. and *Phyllosticta digitalis* Bell, respectively. The first disease was of the anthracnose type, and was found only on young plants, the others were leaf-spots which developed later in the season on established plants. This paper is concerned with the symptomatology of these diseases and the morphology and taxonomy of the parasites, especially with that of *Colletotrichum fuscum* which is most probably new to Great Britain.

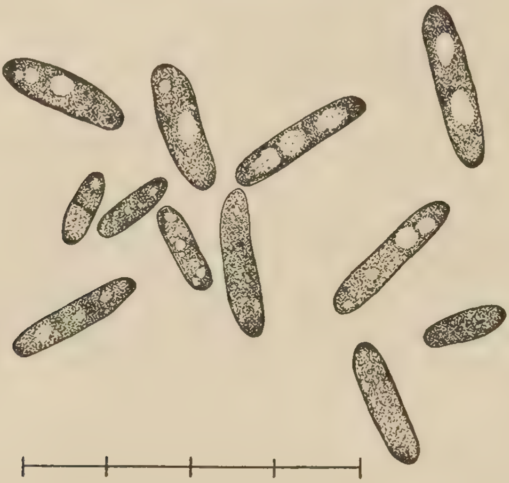
ANTHRACNOSE (*COLLETOTRICHUM FUSCUM* LAUBERT)

Description of the disease and the causal fungus

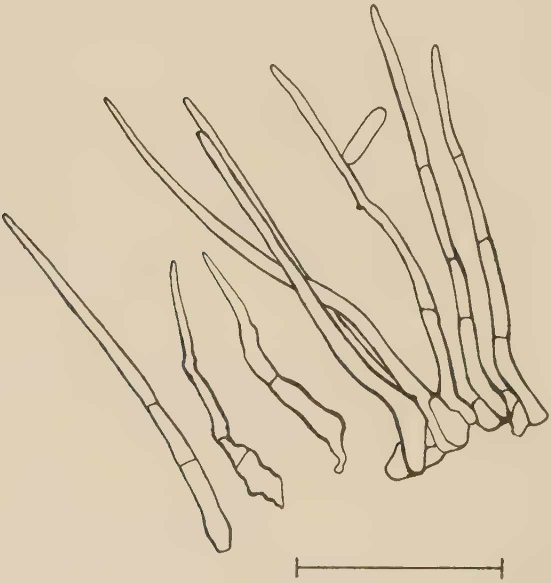
In early July 1952, young plants of *Digitalis lanata* Ehrh. growing at Dartford, Kent, were seen to be affected by a disease characterized by spotting of the leaves, and later typical symptoms of anthracnose. Lower leaves are the first to become infected. Later, the petioles decay and younger leaves become infected until in severe cases the entire aerial portion of the plant becomes blackish-brown and dies. The spots on the leaves (see Pl. 18, fig. 2) are purplish-brown and with increasing age tend to become less dark in colour centrally and attain a maximum diameter of 1·5 mm. Where they are numerous, coalescence occurs and a necrotic area is soon formed in the leaf, but where they are more widely spaced the leaf tissue between them becomes yellowish and then brownish so that large brown patches are ultimately formed. When the disease is in an advanced state the whole leaf, usually commencing at the tip, becomes discoloured and dies.

The infection was first noticed on plants which had been grown from continental seed, but the disease was later found on plants raised from seed from other sources and cross-infection was presumed to have taken place in the field.

Examination of the central portions of maculae revealed, depending on their size, one or many acervuli which in the later stages of the disease were amphigenous but which on younger leaves were mainly hypophyllous. The size of these acervuli, which were very quickly erumpent, was extremely variable, but well-developed ones on living leaves were 220–270μ in diameter. On the younger lesions they were considerably smaller and on dead portions of leaf where the fungal growth was presumably saprophytic rather than parasitic they frequently become confluent, when an estimate of their diameter is probably of little significance. Setae of various lengths are very prominent and around their bases can be seen a large accumulation of discharged conidia. (See Pl. 19, fig. 2.) The setae in this type of acervulus were 80–100 μ in length and contained a variable number of transverse septa. The lower portions of the setae possess thick brownish walls; distally the walls become considerably thinner and the seta usually terminates in a hyaline tip. Many are straight but some are curved and may even be geniculate, and examination of older acervuli or



Text-fig. 1. Conidia of *Colletotrichum fuscum*, showing granular appearance and variable numbers of guttules. 40 μ.



Text-fig. 2. Setae of *Colletotrichum fuscum*. The outline of a spore is included to demonstrate its relative size. The setae on the right are attached to a portion of the sclerotium. 50 μ.

those which have developed on dead tissue shows that the tendency towards septation and malformation is more marked in such examples and that these setae are also markedly shorter than those of normal acervuli. Occasionally very young acervuli and very infrequently larger and older ones do not develop setae (see Pl. 19, fig. 1). The conidia are variable in size and shape and the conidiophores to which they are attached are approximately equal in length or rather shorter than the mature spores which are abstricted from pronounced sterigmata. The group of setae shown in Text-fig. 2 demonstrates the variation which can occur in their appearance. The six on the right were taken from a typical acervulus and the two shorter ones from a leaf showing an advanced stage of infection. The remaining seta is from a very young acervulus in which it was the only specimen present.

Conidia from typical lesions were $14–22 \times 2{\cdot}2–4{\cdot}4\mu$; in older ones greater variation in size was found, but they were all thin-walled, hyaline, cylindric and straight or very slightly curved (see Text-fig. 1). The ends were obtuse or bluntly pointed, one end of the spore frequently being dissimilar to the other and sometimes even slightly subclavate. The spores were invariably granular in appearance but the size and noticeability of the granules varied somewhat with age. Great variation was observed in the numbers of oil droplets in the cells, small conidia frequently possessing none whereas larger ones usually contained at least one and often more, especially when the guttules were small. The occurrence of a solitary central guttule was infrequent as was also the presence of a false septum across the middle of the cell produced by an aggregation of the more opaque cytoplasmic granules.

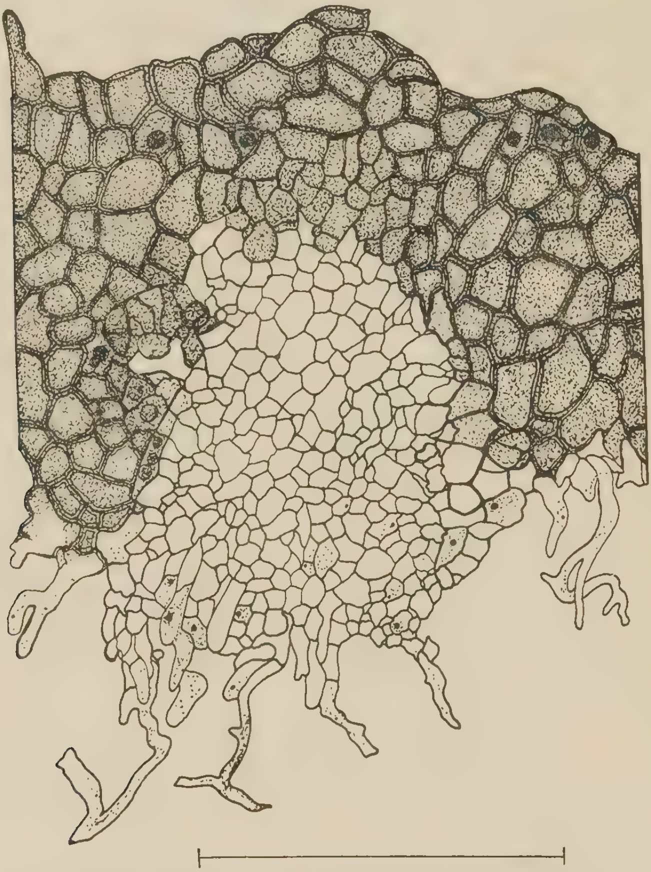
Many of the larger acervuli produced a sclerotium beneath the layer of conidiophores and setae, and when ageing had taken place only the sclerotium and setae remained. On dead portions of the leaf the fungus frequently grew as a sclerotium only, on the surface of which a few stunted setae might be found.

Culture of the fungus

Cultures were readily obtained by plating out suitable dilutions of a suspension in 1/10,000 Calsolene of the numerous spores produced by the younger acervuli. The type of growth obtained after incubation for one week at 23° C. is shown in Pl. 20, fig. 3. On potato-glucose agar the fungus grew rapidly and produced a somewhat floccose growth in which setae could be seen on the older portions of the colony. Similar growth occurred on malt agar but the mycelium did not spread so rapidly. Growth was poorer on oatmeal agar but plentiful sclerotia and setae were formed in the centre of the colony. Sclerotia but no setae were produced on Czapek-Dox agar.

After a fortnight's growth the fungus had completely covered the surface of the agar in all the media used and a continuous black sclerotium was formed on oatmeal and Czapek-Dox media. On the other media the sclerotia remained discrete except for coalescence in the central portions of the colonies. Sporulation did not take place in any of the cultures, neither was there any pigment formed in the mycelium or agar with the exception of the brown or black material colouring the setae and sclerotia.

A section through the sclerotium produced on oatmeal agar (see Text-fig. 3) shows that it is mainly formed around a mass of pseudoparenchyma which lies in contact with the agar and from which separate hyphae penetrate the bulk of the medium. The latter can be seen to contain nuclei and cytoplasm but cell contents are not visible within the pseudoparenchyma. The sclerotium itself consists of blackish, closely aggregated thick-walled cells in some of which oil droplets are visible. Portions of the sclerotium are frequently extremely rich in fatty material and the oil droplets are freed when the tissue is crushed beneath a cover-slip. In old cultures a scanty growth of funiculose mycelium frequently forms above the surface of the sclerotium. The cells of the sclerotium in culture are very similar to those seen in old sclerotia on dead leaves of *Digitalis*.



Text-fig. 3. Longitudinal section through a sclerotium of *Colletotrichum fuscum* developed after a fortnight's growth on oatmeal agar. The material was fixed in La Cour's 2BD and stained with Heidenhain's haematoxylin. 100 μ.

Taxonomy of the causal organism

The above description of the fungus causing anthracnose of *Digitalis lanata* can now be compared with those of similar fungi described in the literature, the salient features of which have been included in Table 1. Two species of *Vermicularia* are described by Grove (1937) as possessing straight or slightly curved spores, *V. herbarum* Westend., and *V. atramentaria* B. et Br., and the latter is considered by him to be synonymous with *V. maculans* Desm., *V. orthospora* Sacc. et Roum., *Colletotrichum solanicolum* O'Gara., *C. tabaficum* Pethybr., *Vermicularia varians* Ducom., and *Colletotrichum lycopersici* Ell. et Ev. Of these only *Vermicularia herbarum* Westend. is reported as occurring on *Digitalis* and the record given is from Ayrshire. *Vermicularia herbarum* f. *dianthi* Westd., is recorded as growing on cultivated carnations and the type is mentioned as also growing on dead stems and leaves of potato. This latter is of significance because this plant is the normal host for *V. atramentaria* over the whole of Great Britain and in many other countries. Grove's description of *V. herbarum* is very similar to that of our fungus except that the acervuli of the latter are rather too small, and rather more in accordance with the size which he describes for *V. atramentaria*. However, the setae of *V. atramentaria* would seem to be longer than those of our fungus, and also its spores are smaller. Grove, quoting Berkley, states that there is sometimes a false septum, protoplasmic in origin, across the conidium of *V. atramentaria* and this is presumably similar in appearance to that shown in Text-fig. 1. With reference to his suggestion that *V. atramentaria* is synonymous with *Colletotrichum lycopersici* Ell. et Ev. it should be emphasized that his description of the setae in the latter fungus is extremely unlike that of the former, and that the sporophores are quite different. *C. lycopersici* is recorded from Worcester, and the distribution of *Vermicularia atramentaria* is given as extending to Europe, North America, South Africa, Australia and India. *V. herbarum* is described by Allescher (1901) as occurring on *Solanum*, *Dianthus*, *Sedum*, *Erythrina* and *Yucca* with a form *armoraciae* on *Cochlearia*. This is an extremely wide host range and it might be considered by some that such a plurivorous species was a *nomen ambiguum*.

In 1928, Duke described a thorough investigation which she made of the British species of *Vermicularia* Fr. and *Colletotrichum* Cda., and concluded that all the species should be transferred to *Colletotrichum* because it was impossible to establish any dividing line between the genera. It is of interest for purposes of comparison with *Vermicularia herbarum* that she recorded *Colletotrichum trichellum* (Fr.) Duke, on an almost equally wide host range; *Magnolia*, *Salix*, *Smilax*, *Arum*, *Pyrus*, *Acer* and *Castanea*, and that despite such an observation she created a new species, *Colletotrichum wahlenbergiae*, for a fungus growing on *Wahlenbergia* merely on a presumed host specificity and a variation of size in morphological elements which was certainly not greater than that which has been indicated above as occurring between measurements made on acervuli obtained from different leaves of the same plant of *Digitalis*. Duke and also Dickson (1926) describe cultures of *Colletotrichum atramentarium* (B. et Br.) Taub., as producing an amethystine pigment in the medium but this has not been

Table 1. *Comparison of* Colletotrichum fuscum *with related fungi*

Fungus and Reference	Acervuli	Conidiophores	Conidia		Setae	
			Size	Shape, etc.	Size	Shape, etc.
Colletotrichum fuscum Laubert Laubert (1937)	200 μ	—	12–24 μ long	Long, straight or somewhat clavate and slightly curved at the narrower end. Containing vacuoles of various sizes	45–100 × 2·4 μ	1–2 septa towards base
Goto's fungus Goto (1938)	Amphigenous, 24–120 μ	Hyaline, rod-shaped tapering towards the tip. Approximately equal in length to conidia	15–21 × 3·5–4 μ (Takumoto) 13–19 × 3·5–4·5 μ	Long, elliptic to cylindric, both ends blunt or sub-acuminate at one end. Straight or slightly curved	100–120 × 4–6 μ at base 74–113 × 3·5–4 μ at base	3–5 septate but only 1–2 easily visible at base. Dark brown, long, often nodular at middle, tapering and hyaline at tip, straight or flexuose
C. digitalis Moesz Goto (1938)	Hypophyllous, confluent, up to 750 μ	—	10–15 × 3 μ	Cylindric with blunt ends	75 × 4–5 μ (7·5 μ at base)	1–2 septate
C. digitalis Unamuno Goto (1938)	Black, gregarious, 88–100 × 25 μ	—	16–22 × 3–3·5 μ	Cylindric, usually straight, sometimes slightly curved	63 × 3·5–4 μ	Brown, septate, strongly curved or flexuose, often nodular
Vermicularia herbarum Westend. Grove (1937)	Black, 300–500 μ	—	18–22 × 3–4 μ	Cylindric or subfusoid-lanceolate nearly always straight, obtuse or somewhat acute at ends. Colourless and granular within, often with minute central guttule	Up to 100 μ	—
C. atramentarium (Grove, 1937) as *V. atramentaria*	Black, 200–350 μ	Short, oblong, brownish	16–22 × 3–4 μ	Linear, straight, obtuse at both ends, even subclavate. Cloudy or granular often with guttule in middle	Up to 200 μ (5 μ broad)	Sparsely septate. Long, brown, straight, somewhat pointed. Surrounded at base by intricate fibres which creep beneath matrix and form large or small velvety violet or black patches
C. atramentarium (Grove, 1937) as *C. lycopersici*	Rusty-brown to black, flat, 90–150 μ	Slender, 30–40 μ long	16–22 × 4 μ	Oblong, ends sub-acute, hyaline 2–3 guttulate	65–112 × 5 μ	Abundant, smoky brown, generally curved, rarely undulate or straight, often geniculate
C. atramentarium Wade (1949)	Black sclerotia, 300–500 μ	—	20–56 × 4–12 μ, mostly 25 × 6 μ	Conidia usually biguttulate but sometimes containing up to 5 oil droplets	—	—
C. biologicum Chaudhuri Chaudhuri (1924)	Black sclerotia, 400–500 × 300–360 μ Young acervuli gregarious, black, 200–300 × 150 μ	Cylindrical, septate 30–60 × 4–6 μ	22 × 5 μ or 14 × 5 μ	Cylindric. Containing several guttules	Up to 130 × 5 μ	Setae simple, black, straight, acuminate, hyaline at apex
C. wahlenbergiae Duke Duke (1928)	—	—	16–22 × 3–4·5 μ	As *C. atramentarium*	Up to 125 μ	Dark brown at base, almost hyaline at tip
Gloeosporium digitalis	—	—	8–10 × 3–4 μ	—	Absent	—

mentioned by all authors as characteristic of the species. Dickson considers that *C. atramentarium* is synonymous with *C. biologicum* Chaudhuri, but the description of this fungus indicates that it produces no pigment. This species is described by Chaudhuri (1924) as producing spores of two different ranges of size and in many cases very few spores or none. This seems to indicate that it is similar to *C. wahlenbergiae*. The production of many oil droplets in the hyphae of cultures of the above species is a character shared by our fungus. In relation to the spores of *C. atramentarium*, Wade (1949) records unusually large spores of this fungus growing on Bismarck potato tubers in Australia. An investigation by Thomas (1939) on the physiology of *C. falcatum* showed that the size of its spores was influenced markedly by the nature of the nitrogen source employed in the culture media and that the fungus made very poor growth when supplied with starch as a carbohydrate source, or with ammoniacal nitrogen. It was unable to utilize cellulose. This may well be an explanation of the different descriptions of *C. atramentarium* which are available and it is also interesting to speculate as to whether it can be considered to be identical with *Vermicularia herbarum*. The question as to whether *Colletotrichum* and *Vermicularia* should be distinguished as separate genera is a rather vexed one. They were considered to be distinct by Grove (1937) who did not believe that *Colletotrichum wahlenbergiae* could possibly be a *Colletotrichum* because it had straight spores. This position was maintained by Wollenweber & Hochapfel (1947) who would retain *Vermicularia* as a genus with fusiform to falcate conidia and an abundance of setae, whereas in *Colletotrichum* the presence of setae is apparently variable and inessential. If this conclusion be adopted there is very little doubt that our fungus must be classified as a *Colletotrichum*, which may be identical with *Vermicularia herbarum*, and has probably some relationship with *Colletotrichum atramentarium*. It may therefore have occurred in this country before the present outbreak of disease.

Investigation on the literature concerning the infection of *Digitalis* in other parts of the world with species of *Colletotrichum* indicates that our disease can probably best be attributed to *C. fuscum* Laubert, but it is somewhat surprising that none of the authors has commented on the similarity between this fungus and *C. atramentarium*. This is probably the result of the identification of pathogenic Fungi Imperfecti from their host without reference to pathogenicity trials aimed at establishing their identity with species of similar or identical morphology.

Colletotrichum fuscum was first described by Laubert (1937) from specimens growing on the leaves of *Digitalis purpurea* L., in a garden in Berlin in August 1927, and an epidemic of the disease was recorded by Thomas (1949) in Pennsylvania. He stated that the fungus was seed-borne and that by means of the fructifications produced on damped-off seedlings it could be rapidly disseminated throughout the majority of plants in the seed-bed. *D. purpurea*, *D. ambigua* and *D. ferruginea* were susceptible to the disease as well as *D. lanata*. Under such conditions it is possible that a residual locus of infection may be established in the soil as described for *Colletotrichum indicum* by Thomas (1939).

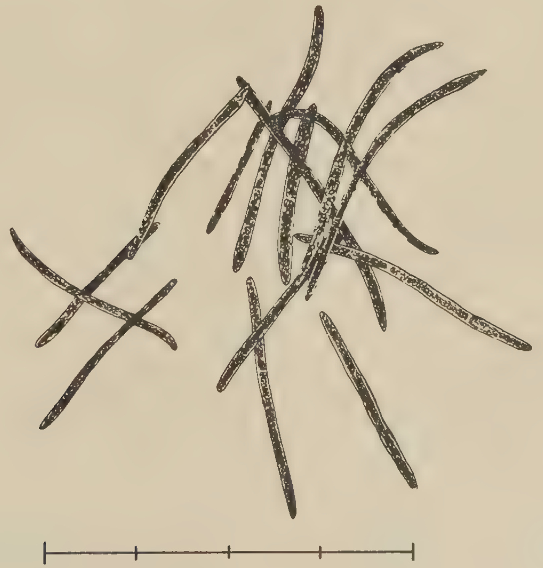
A similar fungus has been described as *C. digitalis* by Unamuno (1933) in Spain and another of which the first name is a later homonym as *C. digitalis* by Moesz (1931). Goto (1938) demonstrated the identity of these two species with *C. fuscum* Laubert. The descriptions of these fungi together with that of *Gloeosporium digitalis* Rostr. are given in Table 1, and with the exception of the latter fungus they evince a remarkable similarity. Moesz (1931) considered that *G. digitalis* was a form of his *Colletotrichum* which was less well developed and lacked the characteristic setae of the larger fungus. The *Gloeosporium* has not been recorded in England and there are grave difficulties in accepting the acervuli lacking setae and pictured in Pl. 19, fig. 1, as belonging to this fungus, so that at present it is better to regard this species as distinct. Its much smaller spore is almost sufficient to distinguish it from *Colletotrichum* without further evidence. Should the three species of *Colletotrichum* cited above prove to be identical, the valid name is *C. digitalis* Moesz, but Goto (1938) could not be certain of this and preferred to regard the fungi which he studied in Japan as being the same as *C. fuscum* and the evidence at present suggests that, as our fungus is extremely similar to the Japanese one, it is probably better to employ this name for the present. Goto (1938) stated that Takumoto, working at the Imperial University of Kyusu, ascertained that his—Goto's—fungus could not infect *Antirrhinum* but no details of attempts to infect plants susceptible to *Colletotrichum atramentarium* and *Vermicularia herbarum* are available and such will obviously need to be undertaken before the status of *Colletotrichum fuscum* can properly be determined.

POSSIBLE IMPORTANCE OF ANTHRACNOSE AND SUGGESTIONS FOR CONTROL

Colletotrichum fuscum seems to be widespread throughout Europe. In addition to the records mentioned above it has been identified at Pavia (Ciferri, 1944) on both *Digitalis lanata* and *D. purpurea* and the great havoc wrought in Pennsylvania by the fungus shows that it should not be regarded lightly as a threat to the cultivation of *D. lanata*. The possibility of the fungus entering the seed is a serious one and it seems advisable to model control measures on those used for *Colletotrichum lindemuthianum* (Sacc. et Magn.) Bri. et Cav. which is responsible for bean anthracnose. The first emphasis is to be laid on the production of seed in disease-free areas and on the mycological examination of seed before sowing, because seed-disinfection has been shown to be ineffective in controlling *Colletotrichum*, and it is also important to remove infected plants from the field as soon as the symptoms have been confirmed. The dispersal of *Colletotrichum* species is generally effected by the splashing of rain drops, and heavy rain in combination with favourable conditions of temperature will greatly increase the rapidity of spread of the disease. In such cases the use of Bordeaux mixture or some other copper fungicide would probably be satisfactory, but nothing is at present known concerning the precise effect of such treatment. *C. lindemuthianum* is most troublesome during the early summer, for the spring, although sufficiently damp, is considerably below the optimum growth temperature of the pathogen whereas the late summer is generally unfavourable because growth is limited both by the lack of moisture and too high a temperature. Such limitations of growth probably apply equally to *C. fuscum* although it should not be assumed that the climatic influences may be necessarily either so specific or so effective as in the example of bean anthracnose. The growing of other species of *Digitalis* near *D. lanata* should be avoided because, should these become infected, their light seeds could easily become carried to the *D. lanata* plants, and the *Colletotrichum* would then readily infect the cultivated crop.

SEPTORIA DIGITALIS PASS. LEAF SPOT

This disease was first noticed on some *Digitalis lanata* plants growing in pots in a cold-frame at Dartford which were being raised for experimental purposes. It was later found to be producing a fairly extensive infestation of plants in a nearby experimental plot and a less severe one on other plots in the same locality. Examination of large production plots in Kent, revealed several specimens of the disease and one diseased plant has been noticed in the field at Beckenham. Dr Ir. G. Elzenga of the Instituut voor de veredeling van tuinbouwgewassen, Wageningen, Holland, states (*in litt.*) that a sporadic infection of *D. lanata* with this fungus occurred in 1952, the year in which the above records were made, and that the disease was first recorded in that country in 1951, when the attack was more widespread than during 1952.



Text-fig. 4. Spores of *Septoria digitalis* Pass. as seen after mounting in cotton blue in lactophenol. 40μ.

In the specimens from Kent the spots are quite large but in specimens which have been received from Holland they are considerably smaller and their size seems to be inversely correlated with the number of original loci of infection. The macula is rather irregular in shape and fuscous with a purple border (Pl. 18, fig. 1) and the pycnidia are minute and scattered over its central area which is generally pallid. The spores are filiform (Text-fig. 4) and correspond in size to those described by Grove (1935), which were found on *D. purpurea* in Cheshire. In the latter specimen Grove also discovered some oblong, curvulous spores $15–20 \times 2{\cdot}5–3{\cdot}0\,\mu$ which he attributed to *Ascochyta digitalis* Fuckel. Spores of a size similar to these were found in specimens from Spain by Unamuno (1933) who described a new species, *Septoria digitalicola*. It seems likely that all these may be growth forms of the same fungus but no spores possessing such measurements have been found in our specimens.

This fungus has a wide distribution. In addition to the records already noted, Allescher (1901) records it from Northern Italy and Lindquist (1941) from South America.

The pycnidia (see Pl. 20, fig. 1) are comparatively thick walled and would probably persist for a considerable time, even after the dead leaf had dropped from the plant. In the presence of moisture the contents swell and the spores are discharged from the central ostiole. They are then conveyed to adjacent leaves by splashing, and will also travel through the surface film of moisture to set up new infections on the same leaf. Rain drops are probably the most efficacious means of dispersal but dry spores might readily be carried about by the wind. Infection of leaves seems to occur irrespective of their age, in contradistinction to the situation found with *Colletotrichum fuscum*. Dr Elzenga has informed me that copper-containing fungicides, applied as sprays, have been effective in checking the spread of the disease in Holland but that repeated applications are necessary to achieve complete success. Preventive measures would therefore seem to be, as usual, more effective than treatment of an established infection.

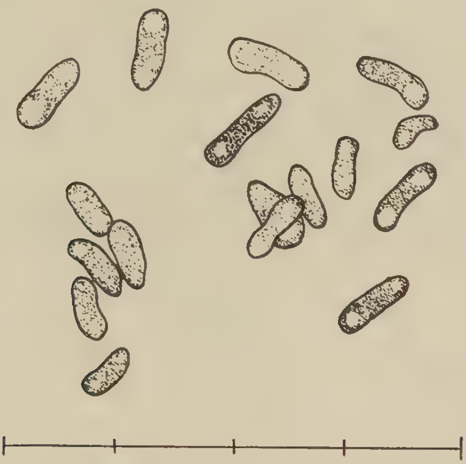
PHYLLOSTICTA DIGITALIS BELL LEAF SPOT

A possible alternative host for the *Septoria* leaf spot was sought at Dartford, and plants of *Digitalis purpurea* growing there were examined but no trace of the fungus could be found on them. However, it was noticed that the lower leaves of adult plants were in an unhealthy condition resulting from attacks by aphides and slugs, and on these leaves were found pycnidia of *Phyllosticta digitalis* Bell (see Pl. 18, fig. 3). Differentiation between *Phyllosticta* and *Septoria* is facilitated by microscopic examination. It will be seen that the spores of the former (Text-fig. 5), which were drawn from a specimen on *Digitalis lanata*, are completely dissimilar and that the pycnidia have a much thinner wall and an eccentric ostiole (Pl. 20, fig. 2). The spores average $7 \times 2{\cdot}5\,\mu$ and are usually biguttulate. These features agree with the published descriptions of *Phyllosticta digitalis* Bell, which has been recorded in England by Grove (1935) and in Belgium and Switzerland by Allescher (1901). The fungus first appeared in Canada in 1943 on several species of *Digitalis* according to Conners & Savile (1944).

On *D. purpurea* at Dartford the pycnidia developed anywhere in the vein islets; but in *D. lanata*, in only a few plants of which species infections by *Phyllosticta* were discovered, it was noticed that they were chiefly restricted to the areas adjacent to the veins in the middle of the leaf.

Furthermore, they commonly occur near the base of the leaf and the resultant necrosis produces a brownish tract of discoloration extending distally from the petiole.

Under moist conditions the contents of the pycnidia swell and the spores are extruded through the ostioles in a vermiform mass. The spores adhere and the spore mass, provided it be untangled, is visible to the naked eye and may be as much as 1 cm. in length. In the presence of excess water the mass breaks up and the spores are liberated as a very thick suspension.



Text-fig. 5. Spores of *Phyllosticta digitalis* Bell as seen after mounting in lactophenol. Note the biguttulate structure of the spores. 40 μ.

Evidence is accumulating that the spores of resupinate Aphyllophorales and other fungi, which inhabit sheltered situations in which neither wind nor water is capable of playing an adequate part in dispersal, are largely zoochorous (Talbot, 1952); and it may well be that the Sphaeropsidales, which produce adhesive spores, rely on a similar mechanism. It would be especially likely that this might occur in those growing near the bases of leaves of rosette plants. It is therefore desirable from the standpoint of controlling *Phyllosticta* that conditions in the field should not be such as to harbour a large population of the molluscs and other invertebrates which normally reside beneath leafy plants.

REFERENCES

Allescher, A. (1901). *Rabenhorst. Kryptogamenflora von Deutschland, Oesterreich und der Schweitz.* Aufl. 2, Pilze, Abt. vi, Leipzig.

Allescher, A. (1903). *Rabenhorst. Kryptogamenflora von Deutschland, Oesterreich und der Schweitz.* Aufl. 2, Pilze, Abt. vii, Leipzig.

Chaudhuri, H. (1924). A new species of *Colletotrichum. Ann. Bot. Lond.* **38**, 735–744.

Ciferri, R. (1944). Relazione sull' attività del Laboratorio Crittogamico dell' Osservatorio Fitopatologico e del Centro Studi Anticrittogamica durante l' anno 1943. *Atti. Ist. bot. Univ. Pavia*, Ser. 5, **1**, 279–632.

Conners, I. L. & Savile, D. B. O. (1944). *Twenty-third Annual Report of the Canadian Plant Disease Survey* 1943.

Dickson, B. T. (1926). The 'Black Dot' disease of potato. *Phytopathology*, **16**, 23–40.

DUKE, M. M. (1928). The genera *Vermicularia* Fr. and *Colletotrichum* Cda. *Trans. Brit. mycol. Soc.* **13**, 156–183.

GOTO, K. (1938). Anthracnose of *Digitalis* caused by *Colletotrichum fuscum*. *Ann. phytopath. Soc. Japan*, **8**, 1–8.

GROVE, W. B. (1935). *British Stem and Leaf Fungi*, **1**. Cambridge.

GROVE, W. B. (1937). *British Stem and Leaf Fungi*, **2**. Cambridge.

LAUBERT, R. (1937). Ein neuer Digitalis-Schädling. *Gartenwelt*, **31**, 674–675.

LINDQUIST, J. C. (1941). Micromicetos nuevos para la flora argentina. *Darwiniana, B. Aires*, **5**, 241–247.

MOESZ, G. (1931). Mykologiai közlemények VIII Közlemény. *Bot. Közl.* **28**, 161–174.

TALBOT, P. H. B. (1952). Dispersal of fungus spores by small animals inhabiting wood and bark. *Trans. Brit. mycol. Soc.* **35**, 123–128.

THOMAS, C. A. (1949). Observations on diseases of some special crops. *Plant Dis. Reptr.* **33**, 453–454.

THOMAS, K. M. (1939). *Detailed administration report of the Government Mycologist, Madras, for the year 1938–1939.* Madras.

UNAMUNO, L. M. (1933). Contribución al estudio de los hongos microscopicos de Galicia. *Rev. Acad. Madr.* **30**, 460–518.

WADE, G. C. (1949). An unusual potato rot. *J. Aust. Inst. agric. Sci.* **15**, 42–43.

WOLLENWEBER, H. W. & HOCHAPFEL, H. (1947). Beiträge zur Kenntnis parasitärer und saprophytischer Pilze, VI, *Vermicularia*, *Colletotrichum*, *Gloeosporium*, *Glomerella* und ihre Beziehung zur Fruchtfaüle. *Z. Parasitenk.* **14**, 181–268.

EXPLANATION OF PLATES 18–20

PLATE 18

Fig. 1. Two plants of *Digitalis lanata* showing heavy infection by *Septoria digitalis*. × 1/4.

Fig. 2. Young leaves of *Digitalis lanata* showing lesions produced by *Colletotrichum fuscum*. × 1 2/5.

Fig. 3. Leaf of *Digitalis purpurea* infected with *Phyllosticta digitalis*. Insect damage is also visible. × 3/4.

PLATE 19

Fig. 1. Acervulus of *Colletotrichum fuscum* devoid of setae, showing the production of spores. × 770.

Fig. 2. Surface view of a well developed acervulus of *Colletotrichum fuscum*. Note the numerous dark setae. × 375.

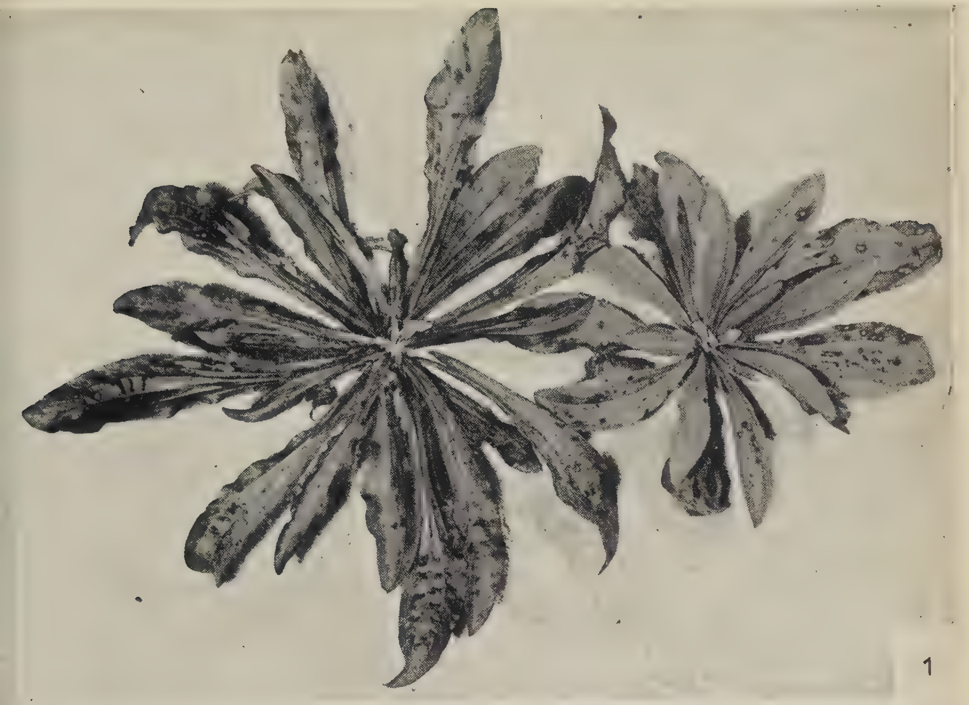
PLATE 20

Fig. 1. *Septoria digitalis*. Surface view of two pycnidia mounted in erythrosin glycerine, showing the large central ostiole. Several conidia are just visible. × 300.

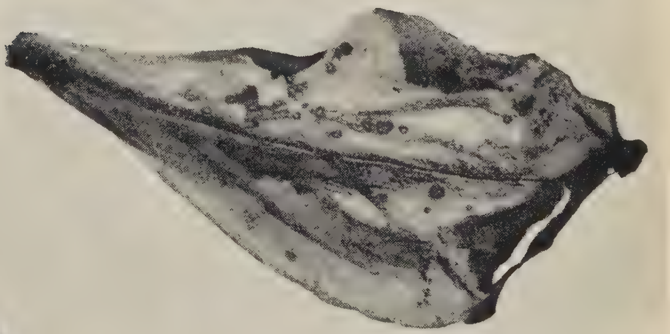
Fig. 2. *Phyllosticta digitalis*. Surface view of four pycnidia mounted in cotton blue in lactophenol. Note that they are less symmetrical than those of Fig. 1 and have much thinner walls. × 300.

Fig. 3. Cultures of *Colletotrichum fuscum*. Top left, oatmeal agar. Top right, Czapek-Dox agar. Bottom left, potato-glucose agar. Bottom right, malt agar. × 5/9.

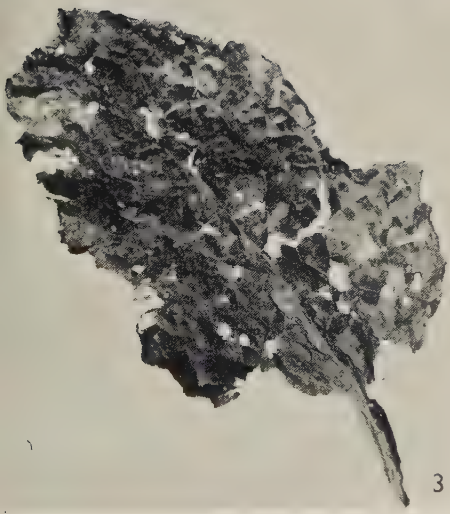
(*Accepted for publication* 20 *November* 1952)



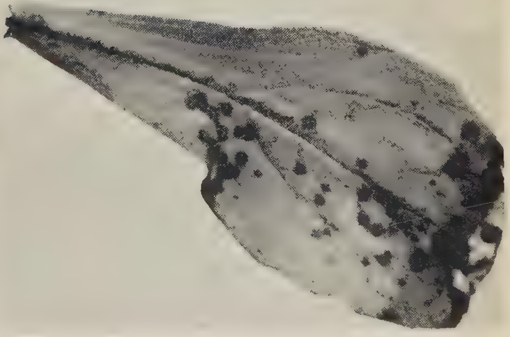
1

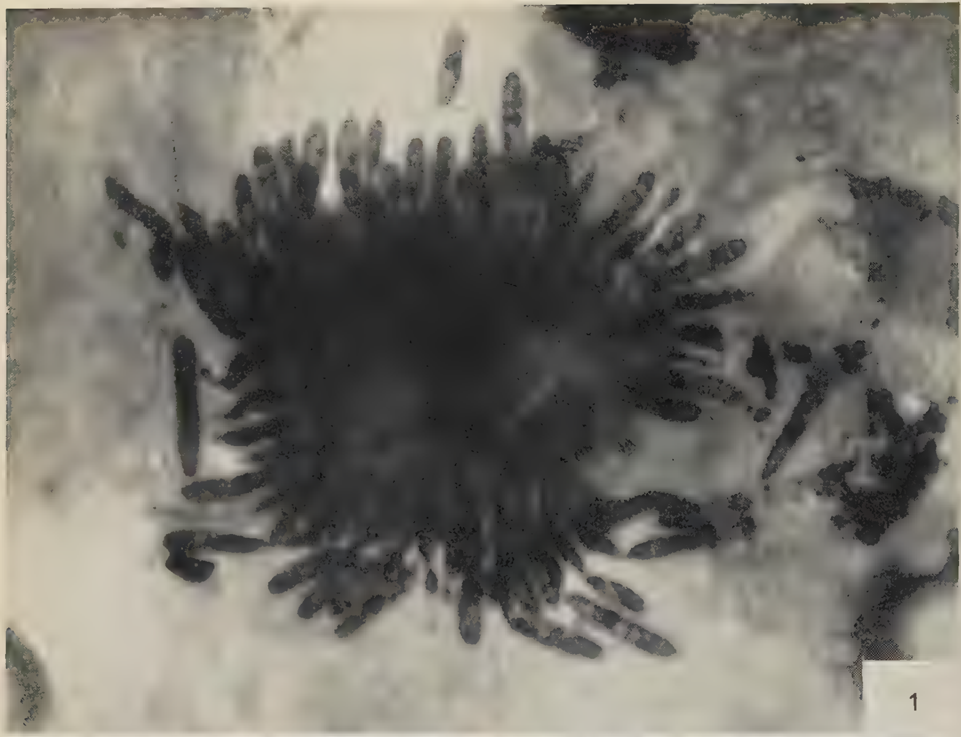


2

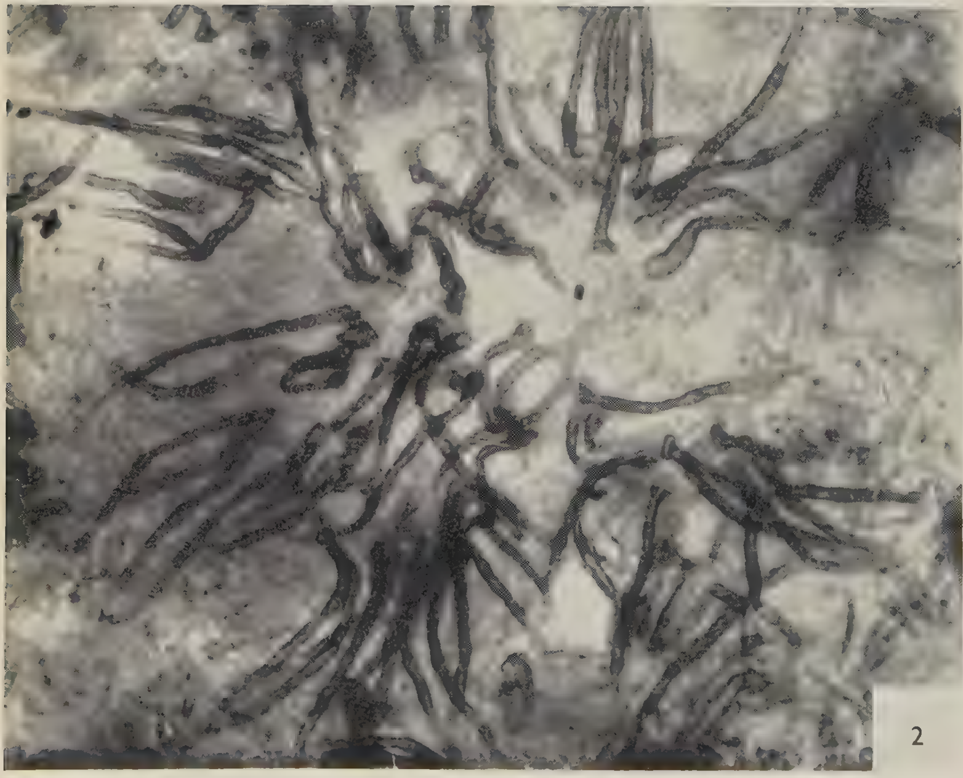


3

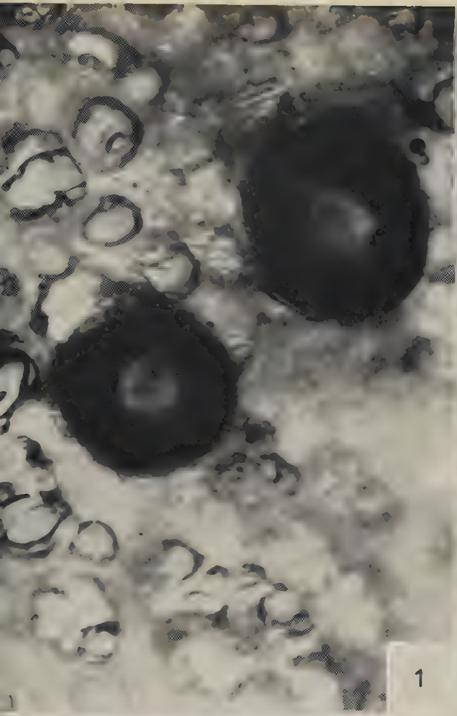




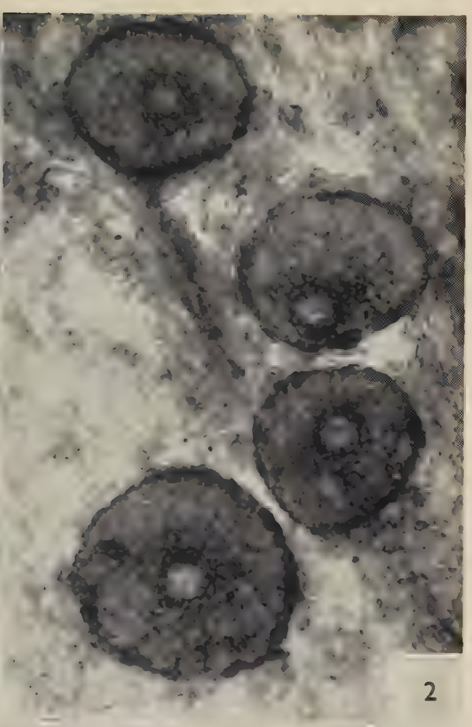
1



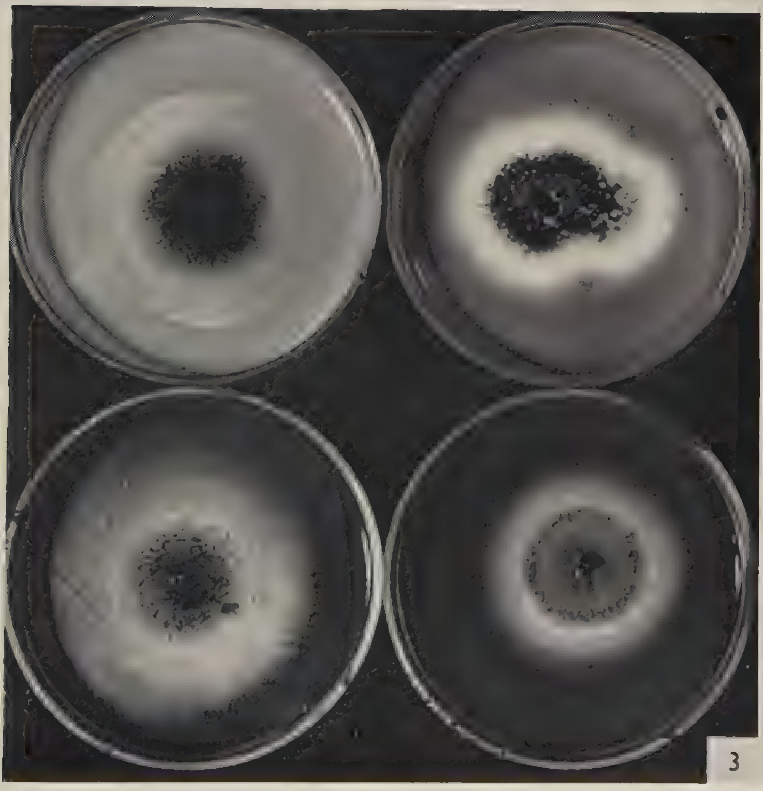
2



1



2



3

OBSERVATIONS ON CHYTRIDIACEOUS PARASITES OF PHANEROGAMS

IV. *PHYSODERMA APONOGETONIS* SP.NOV. PARASITIC ON *APONOGETON**

By F. K. SPARROW

Botany Department, University of Michigan

(With 1 Text-figure)

This paper describes a new species of *Physoderma* parasitic on *Aponogeton undulatus* from Dorset, England. The fungus was undoubtedly present in material of the host imported by air from Ceylon by an aquatic nursery firm. Details of its complete life history are as yet lacking.

Marsh and submerged aquatic phanerogams are not, as might be expected from their habitats, unduly beset with phycomycetous parasites. In the genus *Physoderma* we do have, however, sixteen or more different species which attack these plants. It is the purpose of this paper to record the presence of another species of this genus on the submerged aquatic *Aponogeton undulatus*.

The diseased material of *Aponogeton* was kindly sent me by Dr R. W. G. Dennis, The Herbarium, Royal Botanic Gardens, Kew, who states that it was sent him by a firm of nurserymen in Dorset. They, in turn, had imported the material by air from Ceylon, six weeks previously. Although it cannot be said with certainty just what was the ultimate source of the parasite, it seems very likely that it was on its present host plant when the latter was introduced into the country. A search for it in freshly arrived material of *Aponogeton* might settle this point.

The external manifestation of the fungus on the leaves and petioles is the presence of dark brown irregular patches and streaks. Unlike certain other species, there is no gall formation or other hypertrophy. A search through these brown areas reveals the presence of numerous ellipsoidal resting spores with the distinctive flattening of the lower surface and circumscissile ring of dehiscence. Further, in the greener portions of the leaf the delicate rhizoidal system and numerous septate turbinate cells may be found. These features mark the fungus unmistakably as a species of *Physoderma*. It is hoped that germination of the resting spores and the formation of ephemeral sporangia from the zoospores from resting spores may eventually be accomplished.

Since what little experimental evidence there is seems to point to host specificity in *Physoderma*, it has seemed best to distinguish this form on *Aponogeton* as *Physoderma aponogetonis* sp.nov. (Fig. 1).

---

* Paper No. 975, Botany Department, University of Michigan.

Physoderma aponogetonis sp.nov.

Systema endobioticum ex rhizoideis ramosissimis constans, plerumque tenuissimis sed interdum crassioribus 2 μ diametientibus etiamque cellulis uniseptatis vel biseptatis 12·5–18·7 μ longis, 5–6·2 μ crassis. Sporae perdurantes e latere visae ellipsoidales subtus applanatae, dehiscentes per lineam circumscriptionis, circulares, membrano pallide brunneo 1·25 μ crasso laevi, intus multiguttulatae et vacuolo centrali magno praeditae, 17·5–23·75 μ latae 16·25–23·75 μ altae. Germinatio et sporangia ephemeralia nondum observata.

Parasiticum in folia *Aponogetonis undulati* maculas irregulares brunnescentes formans. Anglia.

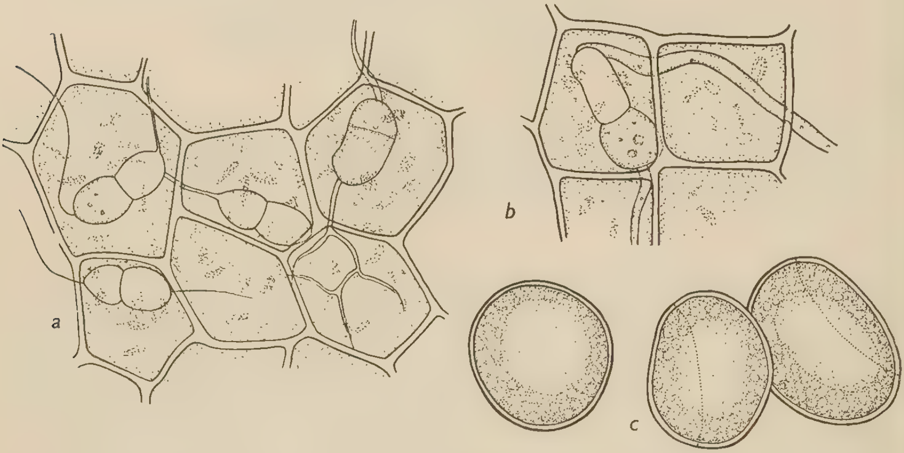


Fig. 1. *Physoderma aponogetonis* sp.nov. *a*, *b*, portions of leaf of *Aponogeton undulatus* showing the endobiotic rhizoidal system bearing turbinate cells; *c*, three mature resting spores. All × 750.

Endobiotic system composed of numerous richly branched rhizoids which are usually of great delicacy although occasionally attaining a diameter of 2 μ, as well as 1–2 septate turbinate cells 12·5–18·7 μ long × 5–6·2 μ in diameter. Resting spores ellipsoidal and flattened on lower side, circular in outline from above, with a circumscissile line of dehiscence, a pale brown smooth wall 1·25 μ thick and guttulate contents surrounding a large central vacuole; 17·5–23·75 μ broad by 16·25–23·75 μ high; germination not observed. Ephemeral sporangia not observed.

Parasitic on leaves and petioles of *Aponogeton undulatus*, where it causes irregular brownish patches and streaks but no hypertrophy. Dorset, England. (Material recently imported from Ceylon.)

Type material has been deposited in the Herbarium of the Royal Botanic Gardens, Kew, and in my own collection.

(*Accepted for publication* 25 *November* 1952)

THE SPORULATION OF *ALTERNARIA SOLANI* IN CULTURE

By K. M. CHARLTON, B.Sc.*

Imperial Chemical Industries Ltd. (Central Agricultural Control), Hawthorndale Laboratories, Jealott's Hill, Bracknell, Berks.

(With Plate 21)

A method for the production of spores from a strain of *Alternaria solani* (Ellis & Martin) Sorauer, using ultra-violet light is described. Sporulation was also induced, but to a lesser degree, by oxidizing agents, in particular by hydrogen peroxide and ozone.

Large supplies of spores of *Alternaria solani* (Ellis & Martin) Sorauer of uniform pathogenicity to tomatoes were required in these Laboratories for the evaluation of fungicides, by a greenhouse method based on that of McCallan & Wellman (1943), using early blight on tomatoes. *A. solani* spores scantily in pure culture. Rands (1917) found that stimulation of sporulation occurred when a 10–12-day-old potato agar culture was cut up and exposed to sunshine. Wounding of the hyphae in cultures on various media produced the same effect (Kunkel, 1918). Ramsey & Bailey (1930) used ultra-violet light. Exposure for 15–30 min. at a distance of 60 cm. from a quartz mercury lamp, with wavelengths between 2535–2800 A. gave good sporulation of the fungus. Dillon Weston (1936) found ultra-violet light to be unnecessary, intense visible light produced the same effect. The methods of Rands and Kunkel have been used by a number of other workers. Nolla (1927) did not obtain spores, whereas Angell (1929), Pironne (1935) and Weber (1939) were more successful. Klaus (1940) found that maximum sporulation occurred at 26° C. with 100% relative humidity and that although the intensity of irradiation influenced sporulation, the effects of temperature and moisture were more important. This was confirmed by Neergaard (1945). McCallan & Chan (1944) obtained their maximum yields of spores on a 2-week-old potato-dextrose agar culture grown at 20° C., which had been scraped, irradiated at 10 cm. distance from a mercury arc lamp emitting about 2500 Å. for 20 sec. and then incubated for 2 days at 20° C.

None of these methods gave yields of spores, from our strain of *A. solani*, sufficiently great to meet our requirements. The present study was made to define a set of conditions from which a standard and reliable method could be established. The effect of oxidizing agents as well as ultra-violet light on the fungus was investigated.

---

* Now at 'Four Oaks', Binfield, Berks.

EXPERIMENTAL

Fungus. Two strains of *A. solani* were used. These were isolated by Dr M. Keay of the Commonwealth Bureau of Plant Breeding and Genetics, Cambridge, in September 1946 from green tomato fruits. They were numbered C.P.B.G. 7 and 14. The former, which is pathogenic to tomatoes and potatoes, was found to spore the more readily and results for this strain only are given.

Culture methods. The cultures were grown in 9 cm. Petri dishes containing 20 ml. of medium. After incubation for 10 days, they were subjected to the treatments described later and incubated for a further 18–24 hr. The spores were then removed by flooding the mycelium with water and rubbing with a bent glass rod. The suspension of spores was then filtered through sandfly netting to remove any pieces of agar and mycelium present, and the concentration of spores counted, using a haemocytometer slide with the Neubauer ruling. A mean of ten counts was taken.

Medium. Ten media were used: malt, malt and maize meal, potato-dextrose, potato-sucrose, Czapek-Dox (sucrose), Czapek-Dox (dextrose), corn steep liquor, glycerol-molasses-peptone and tinned tomato juice, all solidified with agar, potato slices and a liquid potato dextrose medium. Marmite was added to some of the media.

The highest yields of spores were obtained on the Czapek-Dox (sucrose) agar medium containing Marmite. In no instance were spores formed on a liquid medium. The addition of Marmite appeared to induce bifurcation of the beak of the spore.

pH *of medium.* The solid media were examined at pH 5·5, 6·5, 7·0, 8·0. pH 7·0 was the most favourable for spore formation. Cultures often produced an orange-red pigment. It is stated by Bonde (1929) that the formation of the red pigment does not occur with heavily sporing strains of *A. solani*. The pigment is an indicator of pH, turning from yellow on the acid side to a deep red on the alkaline side of pH 7·5. We therefore concluded that the red pigmentation is not a characteristic of a non-sporing strain but rather an indication that the pH of the substrate is above that favourable for abundant sporulation of the fungus.

Temperature. Cultures were grown at 20 ± 1° C. and at 24 ± 1° C. but the temperature range of 19–25° C. was not found to be critical. With a malt medium, 20 ± 1° C. gave a slightly higher yield but 24 ± 1° C. was more favourable with the Czapek-Dox (sucrose) medium.

Type of inoculum. Either a disk of mycelium was placed at the centre of the dish or a suspension in sterile distilled water of spores or mycelium or both was flooded over the surface of the medium. The type of inoculum did not influence the final yield of spores from a culture. The colony resulting from inoculating with a mycelial disk was the most practical method, since a discrete colony was formed, contaminating organisms could readily be recognized and the medium was not exhausted before the spores had been produced. Such a colony could be used to produce as many as 9 crops of spores.

Ultra-violet irradiation. Various doses of ultra-violet light, from an Hanovia fluorescent type X 1 lamp, with and without the Wood's glass filter, were given by varying the time of exposure and the distance of the culture from the arc.

In all our experiments it was found that the irradiation of the cultures, in a manner similar to that used by Ramsey & Bailey (1930) and McCallan & Chan (1944), was the essential process in the initiation of sporulation. Spores were not obtained after exposure of the culture to intense white light. The effective dose of ultra-violet light was 20 min. at a distance of 45 cm. This period of illumination was increased as the lamp aged. It was found preferable to place the cultures in the dark after irradiating as light encouraged the growth of aerial mycelium and made more difficult the removal of the spores. Within a few hours of irradiating, the conidiophores were visible and after 18–24 hr. the spores were mature.

Wounding of mycelium. Some of the cultures were heavily scraped with a scalpel, some flattened with a bent glass rod and some left unscraped prior to exposing to ultra-violet light. Spores were produced under all conditions but due to the presence of aerial mycelium in the unscraped cultures, their removal was difficult.

Water content of the culture. Cultures were dried with filter paper or left with a small amount of surface moisture for the treatment with ultra-violet light. After treatment the cultures were left dry or were flooded with sterile distilled water and then drained. A film of water over the mycelium appeared to act as a partial barrier to the ultra-violet light and there was a decrease in the number of spores produced. After irradiation a high relative humidity was favourable to spore production but the amount of water vapour was in no way critical; there was sufficient water within the medium to supply the requirements of the fungus.

Chemical treatment of the culture. The actual role of the ultra-violet light was a matter of considerable interest. We were of the opinion that the stimulation of sporulation was brought about by an oxidative process in the substrate or mycelium, or both. The site of this process was established by growing cultures on a permeable cellulose film ('Cellophane, permeable 600') on Czapek-Dox (sucrose) agar. After the colonies had grown for a week they were removed from the surface of the agar, irradiated and then replaced on irradiated or non-irradiated media. Non-irradiated cultures were transferred to irradiated and non-irradiated media. It was found that the change occurred within the mycelium and not in the substrate. To test the hypothesis that it was in fact an oxidative process which initiated the formation of spores, cultures were treated with oxidizing and other agents. Cultures, which had the aerial mycelium flattened, were placed in a desiccator and flushed with oxygen and with ozone. No measurements of the concentration were made. Other cultures were washed with solutions of varying concentrations of hydrogen peroxide, potassium permanganate, sodium hypochlorite, potassium persulphate, zinc sulphate and sodium nitrate. After treatment, the cultures were dried with filter paper and after being incubated at 24 ± 1° C. for 18 hr., counts of the spores formed were made. The results are given in Table 1. In no case was the yield of spores as high as that produced by the ultra-violet light treatment but treatment with hydrogen peroxide or ozone produced a moderate degree of sporulation. The result of

Richards (1949) that ozone can induce sporulation was confirmed, but contrary to his finding, the spores we produced were viable and pathogenic to tomato plants.

Table 1. *Effect of chemical agents and ultra-violet light on the sporulation of* A. solani

Agent	Details of treatment	No. of spores per culture
Ultra-violet light	30 min. exposure at a distance of 45 cm. from arc tube	$3{,}160 \times 10^3$
Hydrogen peroxide	1 : 100 H_2O_2 (40 volumes), 5 min. exposure	412×10^3
Ozone	10 min. exposure, concentration not measured	334×10^3
Oxygen	10 min. exposure, concentration not measured	2×10^3
Potassium permanganate	0·1 %, 5 min. exposure	1×10^3
Sodium hypochlorite	0·05 % (concentration near toxic level)	A few isolated spores
Potassium persulphate	1·0 %, 5 min. exposure	1×10^3
Zinc sulphate	1·0 %, 5 min. exposure	1×10^3
Sodium nitrate	2·0 %, 5 min. exposure	1×10^3
Water	—	None

Since ozone can itself induce sporulation and was produced during the operation of the ultra-violet lamp, a barrier to ozone, impermeable 'Cellophane', which allowed ultra-violet through, was placed over a culture during irradiation. Although no ozone reached the culture from the lamp, spores were produced. It was still possible that ozone was formed by the ultra-violet light within the mycelium.

According to Bacq (1951) ultra-violet light may liberate peroxides in the mycelium, hence the part played by peroxides was considered further. It was clear that the mycelium contained the enzyme catalase since it decomposed hydrogen peroxide with the formation of molecular oxygen. But molecular oxygen is not active in promoting sporulation. If peroxidases were also present, the oxygen would not be liberated from hydrogen peroxide in molecular form but would oxidize a substrate.

An external source of peroxidases, an extract of horse-radish, was added to cultures together with hydrogen peroxide. There was no appreciable increase or decrease in the number of spores.

Attempts were made to neutralize the effects of treatment with hydrogen peroxide by flooding the cultures with reducing agents. Neither cysteine at 400 p.p.m. nor ascorbic acid at 1000 p.p.m. affected the production of spores.

After completing these experiments, our attention was drawn to work on the formation of pycnidia by *Plenodomus fuscomaculans* (Coons, 1916). Coons was able to replace the morphogenic action of light in producing pycnidia by using agents such as hydrogen peroxide, nitric acid, sulphuric acid and potassium dichromate, potassium permanganate, ferric chloride, sulphuric acid and zinc sulphate. We obtained positive results with some of these agents but, as found by Coons, the yield of spores was in no case as high as that obtained with light treatment.

STANDARD METHOD FOR THE PRODUCTION OF SPORES OF *A. SOLANI*

After a consideration of the experimental results, the following method has been adopted as standard procedure and has given satisfactory results for a period of 3½ years.

Fungus. A. solani (C.P.B.G. 7; Hawthorndale culture collection K. 30).

Maintenance of stock cultures. Four stock cultures are maintained on 2 % malt agar slopes and stored at 5° C. The culture is also kept on 2 % malt agar slopes under sterile mineral oil and stored at 5° C.* The stock cultures are subcultured at 6-monthly intervals, hence each year the stock cultures are two generations removed from the original culture. Three of the stock cultures are used to provide working cultures over a period of 6 months, and the fourth to maintain the stock.

Medium. Modified Czapek-Dox agar of the following composition is used. (Quantities are given as percentage weight/volume throughout.) Sodium nitrate, 0·2; potassium dihydrogen phosphate, 0·1; potassium chloride, 0·05; magnesium sulphate, 0·05; ferrous sulphate, 0·001; sucrose, 3·0; Marmite, 1·0. The medium is made up with distilled water and the pH adjusted to 6·8–7·0 with *N*/1 sodium hydroxide and solidified with 2·0 % agar. It is sterilized by autoclaving for 20 min. at 15 lb.

Method of culture. A suitable number of Petri dishes (9 cm.), each containing 20 ml. of medium, is prepared. These are inoculated at the centre of the medium with a small piece of mycelium from a working culture. The cultures are then incubated for 10 days at 24 ± 1° C. in the dark.

Preparation of cultures for irradiation with ultra-violet light. When the colony is about 6–8 cm. in diameter after 7–10 days' incubation (Plate 21, B) the fluffy aerial mycelium is flattened using a sterilized bent glass rod. Occasionally the surface of the colony is leathery. This type of culture may be used for the production of spores but requires scraping with a razor blade, to remove the tough skin, prior to irradiating. Surface moisture if present is removed from the culture with filter paper.

Irradiation of cultures. Irradiation is by an Hanovia fluorescent type X 1 lamp with the Wood's glass filter removed. The dose for each culture is 20 min. at 45 cm. As the efficiency of the mercury vapour arc tube decreases with age, the distance from arc tube to culture is decreased or the period of irradiation extended. Owing to fluctuations in the voltage of the supply of electricity, it was found necessary to insert a voltage stabilizer in the circuit. It is convenient to irradiate four cultures at a time. As a safety precaution irradiation is carried out in a cabinet, constructed of red 'Perspex', enclosing the lamp and cultures. Ozone, formed by the lamp, is removed by drawing a stream of air through the cabinet into a wash bottle containing a solution of sodium nitrite. The cultures are then incubated for 18–24 hr. at 24 ± 1° C.

Removal of spores. The spores are usually produced by the young tissue around the edge of the colony but sometimes, after two or more irradiations, they are produced over the whole of the surface of the colony.

---

* The culture has now been freeze-dried in these Laboratories. The freeze-dried material has retained both its ability to form spores and its pathogenicity to tomatoes for one year. This method may provide an alternative means of storing the stock culture.

Heavily sporing cultures present a dark green velvety appearance (Plate 21, A). The spores are removed by flooding the culture with about 15 ml. distilled water by the method already described and the concentration of spores then adjusted to the required density.

Re-use of cultures. After the removal of the spores the cultures may be dried with filter paper and stored at 5° C. for a period of a week. To produce a further batch of spores the cultures are irradiated again as described above, care being taken to flatten any aerial mycelium which has developed during storage and to remove any surface moisture.

Discussion

The strain of *A. solani* used by us did not respond to the methods of other workers for promoting sporulation of this fungus. A different set of cultural conditions was necessary and these we have defined. The method resembles that of McCallan & Chan (1944) but differences in the composition of the medium, temperature of incubation and method of irradiating the culture have been made. In no case, were our yields of spores per culture as high as those claimed by McCallan & Chan, but the numbers of spores produced fully met our demands. Our details of procedure differ but we agree with McCallan & Chan that exposure of the culture to ultra-violet light was the most important factor in inducing sporulation of the fungus.

The effect of the ultra-violet light may well be an indirect one, as suggested by Bacq (1951), due to the formation of peroxides by the action of the ultra-violet irradiation on the molecules of water. This is supported by our experiments in which hydrogen peroxide and ozone produced moderate sporulation of the fungus. Other oxidizing agents, such as potassium permanganate and potassium persulphate gave a slight degree of sporulation. These results suggest that oxidative processes play an important part in the mechanism by which ultra-violet light exerts its influence. However, since none of the chemical treatments tried approached the efficiency of ultra-violet light, their use is of little practical value in producing large numbers of spores of *A. solani*.

Thanks are due to Miss M. F. Bray for technical assistance in carrying out this work.

REFERENCES

Angell, H. R. (1929). Purple blotch of onion (*Macrosporium porri* Ell.). *J. agric. Res.* **38**, 467–487.

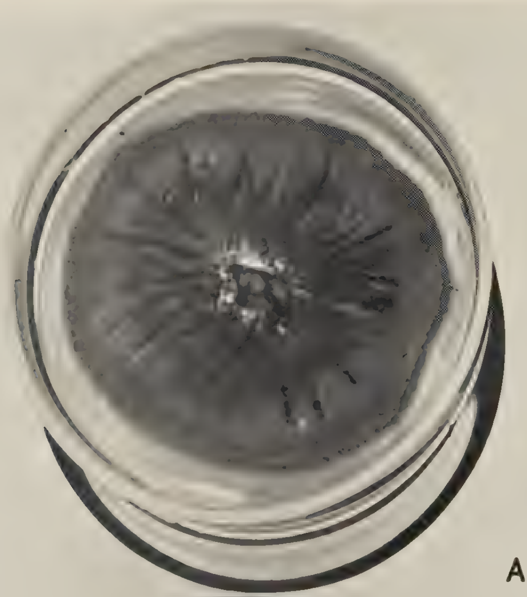
Bacq, Z. M. (1951). L'action indirecte du rayonnement X et ultra-violet. *Experientia*, **7**, 11–19.

Bonde, R. (1929). Physiological strains of *Alternaria solani*. *Phytopathology*, **19**, 6, 533–548.

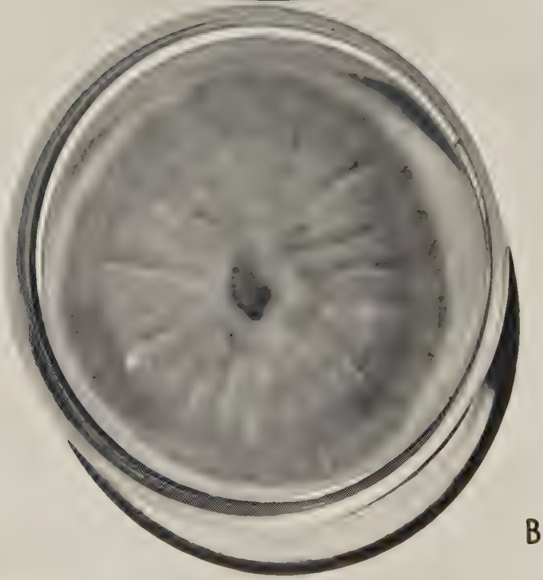
Coons, G. H. (1916). Factors involved in the growth and the pycnidium formation of *Plenodomus fuscomaculans*. *J. agric. Res.* **5**, 713–769.

Dillon Weston, W. A. R. (1936). The sporulation of *Helminthosporium avenae* and *Alternaria solani* in artificial culture. *Trans. Brit. mycol. Soc.* **20**, 112–115.

Klaus, H. (1940). Untersuchungen über *Alternaria solani*, Jones et Grout, ins besondere über seine Pathogenität an Kartoffelknollen in Abhangigkeit von den Aussenfaktoren. *Phytopath. Z.* **13**, 126–195.



A



B

Kunkel, S. O. (1918). A method of obtaining abundant sporulation in cultures of *Macrosporium solani* E. and M. *Brooklyn bot. Gdn. Mem.* **1**, 306–312.

McCallan, S. E. A. & Chan, S. Y. (1944). Inducing sporulation of *Alternaria solani* in culture. *Contr. Boyce Thompson Inst.* **13**, 323–335.

McCallan, S. E. A. & Wellman, R. H. (1943). A greenhouse method of evaluating fungicides by means of tomato foliage diseases. *Contr. Boyce Thompson Inst.* **13**, 93–134.

Neergaard, P. (1945). *Danish species of* Alternia *and* Stemphylium. Copenhagen.

Nolla, J. A. B. (1927). A new *Alternaria* disease of onions (*Allium cepa* L.). *Phytopathology*, **17**, 115–132.

Pironne, P. P. (1935). An improved method for inducing spore fructification in certain species of *Macrosporium.* Abstract in *Phytopathology*, **25**, 31–32.

Ramsey, G. B. & Bailey, A. A. (1930). Effects of ultra-violet radiation upon sporulation in *Macrosporium* and *Fusarium. Bot. Gaz.* **89**, 113–136.

Rands, R. D. (1917). The production of spores by *Alternaria solani* in pure culture. *Phytopathology*, **7**, 316–317.

Richards, M. C. (1949). Abstract in *Phytopathology*, **39**, 20.

Weber, G. F. (1939). Nailhead spot of tomato, caused by *Alternaria tomato* (Cke.) n.comb. *Florida agric. Exp. Sta. Techn. Bull.* **332**, 54.

EXPLANATION OF PLATE 21

Cultures of *Alternaria solani* showing the effect of irradiation on spore production. Cultures were of the same age. Culture A was irradiated 24 hr. before the photograph was taken.

(*Accepted for publication* 27 *November* 1952)

SOME PREDACIOUS FUNGI OCCURRING IN DUNG

By A. J. JUNIPER

Biological Laboratories, The Polytechnic, Regent Street, London

(With Plate 22 and 4 Text-figures)

An account is given of some predacious fungi found in dung, three of them, *Acaulopage acanthospora* Drechsler var. *magna* var.nov., *Acaulopage stenospora* Drechsler, and *Acrostalagmus gonioides* Drechsler, being recorded for the first time in Britain.

Duddington (1951) recorded nine predacious species from animal dungs and of the six species recorded here only one of them, *Arthrobotrys oligospora* Fres., has previously been recorded from this habitat in Britain.

Acaulopage acanthospora Drechsler var. **magna** var.nov.

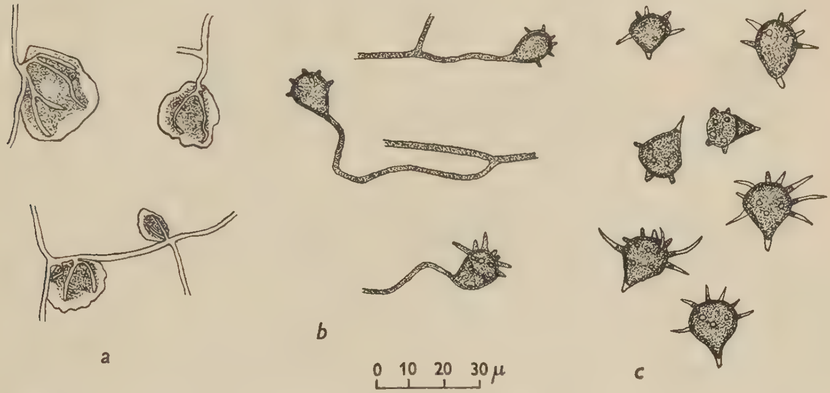
A typo differt conidia 12–19 μ longa, 8–15 μ crassa, 6–16 appendicibus, 5–8 μ longis, 1·5–2 μ crassis ad fundum. ·Hab. in excreta animalis, *Bos taurus* L., Langdon Hills, Essex, Anglia. April 1952.

Type material will be deposited in the Herbarium of the Royal Botanic Gardens, Kew and in the Herbarium of the Commonwealth Mycological Institute, Kew.

Small pieces of cow dung (*Bos taurus* L.), collected from the Open Spaces, Langdon Hills, Essex on 27 April 1952, were placed on sterile maize-meal agar plates the following day; the fungus was first observed capturing amoebae on 26 June 1952.

The mycelium consisted of colourless, branched, non-septate hyphae about 1·5 μ in diameter spreading over the surface of the agar. Amoebae, 5–30 μ in diameter, were observed lying close to the mycelial hyphae; some of these amoebae were captured by the fungus and their protoplasm was absorbed by means of sparingly branched, spreading haustoria. The haustorial branches were about 1 μ in diameter (Text-fig. 1*a*). Conidia developed at the tips of branches of the mycelium (Text-fig. 1*b*). The conidia were hyaline, pear-shaped structures, 12–19 μ long and 8–15 μ wide, bearing 6–16 (commonly 12) appendages at the broader distal end. These appendages were 5–8 μ long and 1·5–2 μ wide at the base; when first formed the appendages were completely filled with cytoplasm, but in mature conidia the distal regions of the appendages were more or less empty (Text-fig. 1*c*; Pl. 22, figs. 1, 2). Sexual reproduction was not observed.

Drechsler has described five species of the genus *Acaulopage* which bear a number of appendages at the distal end of the conidium, namely, *A. tetraceros* (1935), *A. acanthospora* (1938), *A. lasiospora* (1942*a*), *A. hystricospora* (1946) and *A. lophospora* (1946). Of these, *A. tetraceros* has 4–6 long appendages and *A. lasiospora* and *A. hystricospora* have a large number (25–125 and 10–50, respectively) of short distal appendages on the conidium. The fungus I had under observation did not fit into any of these species. The conidial measurements were close to those given by Drechsler for *A. lophospora*, but in this species the conidia are described as being inversely flask-shaped like those of *A. tetraceros*. The conidia of the fungus that I was dealing with were pear-shaped and very like those of *A. acanthospora*, except that they were larger in size and had larger appendages. I have, therefore, described the fungus as a variety of *A. acanthospora*.



Text-fig. 1. *Acaulopage acanthospora* Drechsler var. *magna* var.nov. *a*, portions of mycelium with captured amoebae and haustoria; *b*, developing conidia; *c*, conidia.

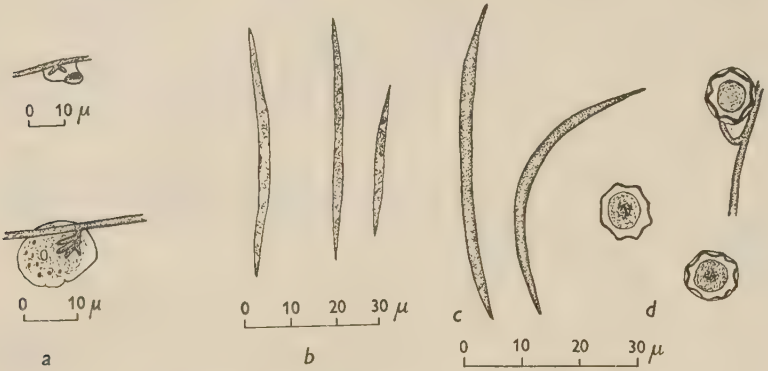
Acaulopage stenospora Drechsler in *Mycologia*, **33**, 257, 1941.

Small pieces of cow dung from a lane at Hainault, Essex, collected on 14 October 1951, were placed on sterile maize-meal agar plates the following day. The fungus was observed near the inoculum 18 days later.

The mycelium consisted of rather poorly branched, non-septate hyphae which were about 1·5 μ wide. Amoebae were seen adhering to the mycelium, and sometimes from the point of contact blunt, pedicellate haustoria penetrated the ectoplasm of the animal. These haustoria branched once or twice and absorbed the cytoplasm of the animal (Text-fig. 2*a*). Conidia developed first in the region of the inoculum and later further out on the agar, appearing, under the low power of the microscope and viewed without a cover glass, as bristle-like projections from the surface of the medium. Each conidium was borne on the tip of a short sterigma. The conidia were elongate, colourless structures, pointed at both ends and either straight or curved (Text-fig. 2*b*, *c*). The cytoplasm was somewhat granular in appearance and the conidia were 23–54 μ long and about 1·5 μ wide.

Zygospores were observed a week after the fungus was first noticed. These were globose, yellowish structures with a thick wall bearing several prominent protuberances. The zygospores were mostly about 7·5 μ in external diameter and mostly 5·5 μ in internal diameter. The zygosporangial wall was sometimes seen collapsed around the zygospore and sometimes the suspensor connecting with the mycelial hypha was observed (Text-fig. 2*d*).

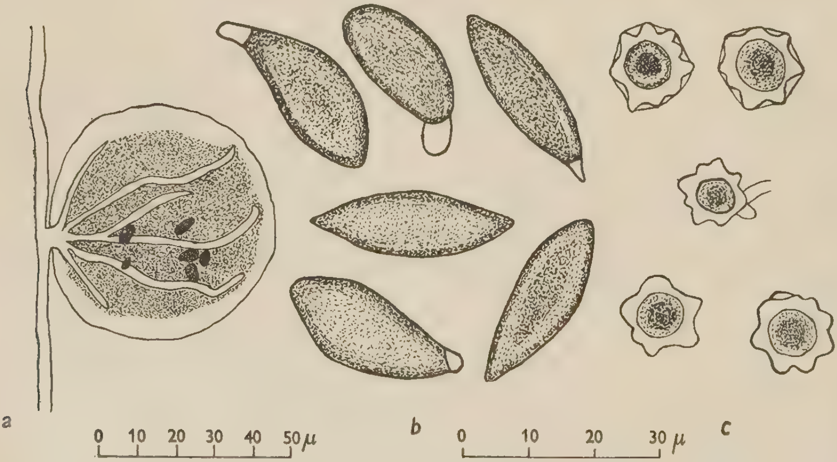
The material I had under examination agreed very closely with that described by Drechsler (1941).



Text-fig. 2. *Acaulopage stenospora* Drechsler. *a*, amoebae with haustoria; *b* and *c*, conidia; *d*, zygospores.

Stylopage rhynchospora Drechsler in *Mycologia*, **31**, 396, 1939.

This fungus was obtained (1) from cow dung collected from a lane at Hainault, Essex, on 14 October 1951 and inoculated on to sterile maize-meal agar plates on 9 January 1952, and (2) from cow dung collected from the Open Spaces, Langdon Hills, Essex, on 25 May 1952 and similarly treated on the following day. In both cases the fungus was first observed about a fortnight after inoculation.



Text-fig. 3. *Stylopage rhynchospora* Drechsler. *a*, amoeba with haustorium; *b*, conidia; *c*, zygospores.

The mycelium consisted of sparingly branched hyphae, about 2 μ in diameter, catching amoebae 50–70 μ in diameter, and absorbing their cytoplasm by means of bush-like haustoria (Text-fig. 3*a*). The fungus produced a large number of erect conidiophores each bearing a single conidium at its tip (Pl. 22, fig. 3). The conidia were 19–38 μ long and 8·5–10 μ wide, ovoid in shape, and usually with an empty beak-like structure at the distal end (Text-fig. 3*b*; Pl. 22, fig. 4).

Zygospores were produced after a period of prolific conidial production. These were yellowish, spherical or subspherical, the wall being rather thick and ornamented with large protuberances, having an external diameter of 11–13 μ and an internal diameter of 6·5–8 μ (Text-fig. 3*c*).

My material agreed with the descriptions of Drechsler (1939) and Duddington (1950).

Stylopage lepte Drechsler in *Mycologia*, **27**, 202, 1935.

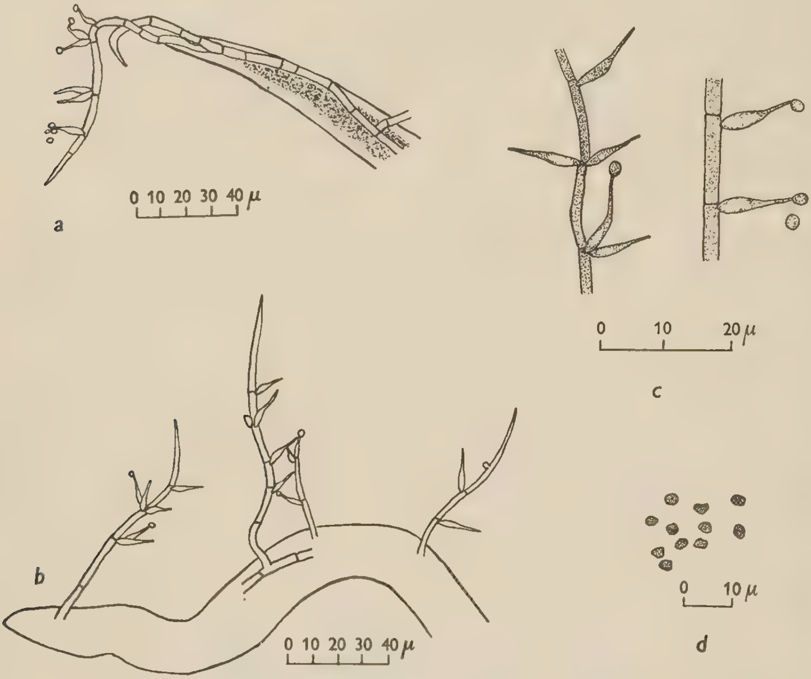
This fungus was isolated from cow dung collected in a lane at Hainault, Essex, on 14 October 1951, appearing on maize-meal agar plates one month after inoculation.

The fungus caught amoebae, devouring their contents by means of slightly branched haustoria. Conidiophores were erect, bearing conidia in alternate arrangement; the conidia being 16–20 μ long and 2–2·5 μ wide. The material I had under observation agreed with that described by Peach (1952) in having up to nine conidia on each conidiophore. Zygospores were not observed.

Acrostalagmus gonioides Drechsler in *J. Wash. Acad. Sci.* **32**, 347, 1942.

This fungus has been isolated from two collections: (1) from cow dung collected from a lane at Hainault, Essex, on 14 October 1951 and inoculated on to sterile maize-meal agar plates on 23 April 1952; and (2) from cow dung collected from the Open Spaces, Langdon Hills, Essex, on 25 May 1952 and similarly treated a month later. In both cases dead nematode worms containing an endozoic fungus were seen on the surface of the agar about a fortnight after inoculation. The integuments of these worms were filled with branched, septate hyphae, 2–3 μ wide, the individual cells being 6–13 μ long. From each nematode several fertile hyphae were produced. These were erect when the worm died on or near the agar surface, but occasionally the worm moved down into the medium before succumbing to the fungus, and the fertile hyphae grew out horizontally into the agar (Text-fig. 4*a*, *b*; Pl. 22, fig. 5). The fertile hyphae were usually 50–300 μ long, septate, 1·5–2 μ wide, with septa 11–16 μ apart. Flask-shaped phialides were developed from the distal ends of the cells of the fertile hyphae, occurring singly or in two's and three's or very occasionally as a group of four (Text-fig. 4*c*; Pl. 22, fig. 6). The phialides were 8–14·5 μ long and 2–3·5 μ in greatest width, terminating in a neck less than 1 μ in diameter and 3–6 μ long. The cytoplasm of the swollen base of the phialide often contained one or two large vacuoles. Adhering to the tip of the phialide was a group of conidia which appeared yellowish when the fertile hypha was aerial and examined without a cover glass. The conidia were more or less polyhedral in shape and were 2–3 μ in diameter (Text-fig. 4*d*).

The material I had under observation agreed with that described by Drechsler (1942*b*) apart from the fact that storage hyphae were not observed, even in cultures 3 months old.



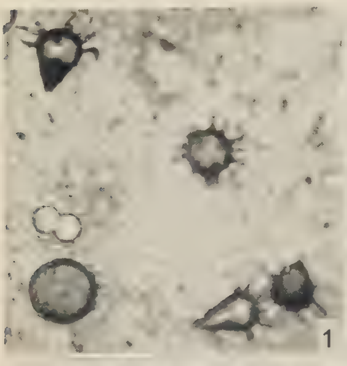
Text-fig. 4. *Acrostalagmus gonioides* Drechsler. *a*, endozoic mycelium and fertile hypha; *b*, portion of nematode with fertile hyphae; *c*, two portions of fertile hypha with phialides; *d*, conidia.

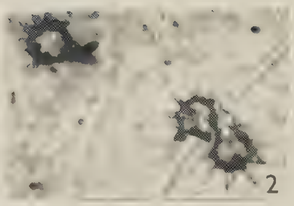
Arthrobotrys oligospora Fresenius in *Beiträge zur Mykologie*, p. 18, 1850.

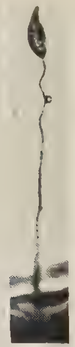
This fungus was isolated from cow dung collected from a lane at Hainault, Essex, on 14 October 1951, and trapped nematode worms by means of three-dimensional networks (Pl. 22, fig. 7). After a short period erect conidiophores were produced bearing whorls of conidia. The conidia were uniseptate, 19–30 μ long and 10–16 μ wide.

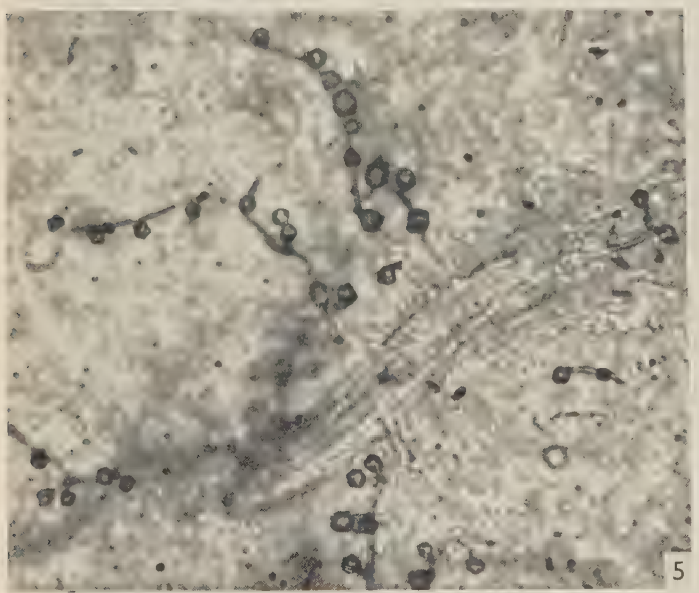
All measurements and drawings used in this paper have been made from living material.

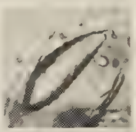
I would like to thank Dr C. L. Duddington for his help and guidance during the course of this work, and Dr R. A. Gillanders for assistance in preparing the Latin diagnosis. My thanks are also due to my colleagues at The Polytechnic for their help and advice.

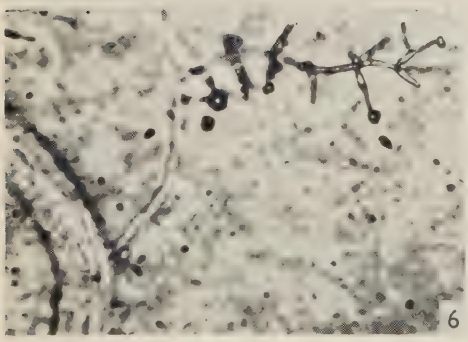

1

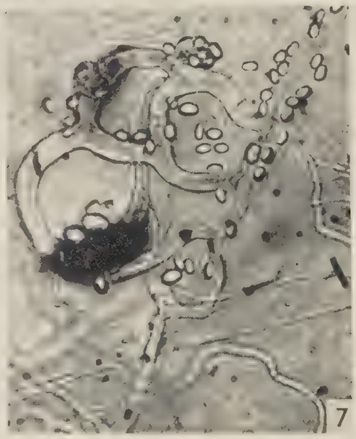

2


3


5


4


6


7

REFERENCES

Drechsler, C. (1935). Some non-catenulate conidial phycomycetes preying on terricolous amoebae. *Mycologia*, **27**, 176–205.

Drechsler, C. (1938). New Zoopagaceae capturing and consuming soil amoebae. *Mycologia*, **30**, 137–157.

Drechsler, C. (1939). Five new Zoopagaceae destructive to rhizopods and nematodes. *Mycologia*, **31**, 388–415.

Drechsler, C. (1941). Four phycomycetes destructive to nematodes and rhizopods. *Mycologia*, **33**, 248–269.

Drechsler, C. (1942*a*). New species of *Acaulopage* and *Cochlonema* destructive to soil amoebae. *Mycologia*, **34**, 274–297.

Drechsler, C. (1942*b*). Two zoophagous species of *Acrostalagmus* with multicellular *Desmidiospora*-like chlamydospores. *J. Wash. Acad. Sci.* **32**, 343–350.

Drechsler, C. (1946). Three new Zoopagaceae subsisting on soil amoebae. *Mycologia*, **38**, 120–143.

Duddington, C. L. (1950). Further records of British predacious fungi. I. *Trans. Brit. mycol. Soc.* **33**, 209–214.

Duddington, C. L. (1951). The ecology of predacious fungi. I. Preliminary survey. *Trans. Brit. mycol. Soc.* **34**, 322–331.

Fresenius, G. (1850). *Beiträge zur Mykologie.* Frankfurt am Main.

Peach, M. (1952). Aquatic predacious fungi. II. *Trans. Brit. mycol. Soc.* **35**, 19–23.

EXPLANATION OF PLATE 22

Figs. 1 and 2. *Acaulopage acanthospora* var. *magna* var.nov., conidia. × 400.

Fig. 3. *Stylopage rhynchospora* Drechsler, conidiophore bearing a terminal conidium. × 180.

Fig. 4. *S. rhynchospora* Drechsler, conidium. × 400.

Fig. 5. *Acrostalagmus gonioides* Drechsler, fertile hyphae growing out from a dead nematode worm with groups of conidia adhering to the tips of the phialides. × 400.

Fig. 6. *A. gonioides* Drechsler, single fertile hypha with phialides. × 400.

Fig. 7. *Arthrobotrys oligospora* Fres., a three-dimensional network. × 400.

(*Accepted for publication* 11 *December* 1952)

STUDIES ON LARCH CANKER

I. THE TAXONOMY AND BIOLOGY OF *TRICHOSCYPHELLA WILLKOMMII* (HART.) NANNF. AND RELATED SPECIES

By J. G. MANNERS

Department of Botany, University of Southampton

(With Plate 23 and 6 Text-figures)

The taxonomy of the larch canker parasite and related fungi is discussed. *Trichoscyphella willkommii* (Hart.) Nannf. is accepted as the name of the parasite and the new combination *T. hahniana* (Seaver) Manners is proposed for the related saprophyte usually known as *Dasyscypha calycina*. *Trichoscyphella willkommii* and *T. hahniana* are shown to be distinct both morphologically and pathologically and a detailed description of each is given. Inoculation experiments carried out in a frost-free locality indicated that *T. willkommii*, but not *T. hahniana*, could cause cankers and die-back on European larch when inoculated into wounds.

Although larch canker has been known for nearly a hundred years, its cause is still in dispute. Most of the earlier investigators, such as Hartig (1880) considered that the disease was caused by the larch canker fungus (*Trichoscyphella willkommii* (Hart.) Nannf.) acting as a wound parasite, and Hiley (1919) held similar views, considering that the fungus caused cankers after entering through dead lateral branches. Day (1931, 1937) considered, however, that cankers were caused by frost, the fungus being merely a secondary invader, while Langner (1936) concluded that both intermittent low temperatures and the presence of the fungus were required. Hahn & Ayers (1943) found that, when artificially inoculated, *T. willkommii* could cause cankers without the intervention of frost. The situation is complicated by the fact that at least two very similar species of *Trichoscyphella* occur on larch and not all investigators have distinguished between them.

In view of the economic importance of larch canker, it seemed desirable that the mode of initiation and persistence of cankers should be fully understood, and hence this investigation was carried out at the suggestion of Mr T. R. Peace (Forestry Commission Research Branch).

THE TAXONOMY OF THE LARGE-SPORED BRITISH SPECIES OF *TRICHOSCYPHELLA*

There is still considerable confusion concerning the number of species of hairy inoperculate Discomycetes occurring on conifers in Britain, and the taxonomy of the species known to occur is in an unsatisfactory condition (Dennis, 1949). These fungi, with massive, stalked apothecia, filamentous paraphyses and non-septate ascospores, form a natural group. Fries (1822) described one of these fungi as *Peziza calycina* Schum. ex Fr. and placed it in Tribe 6 (Dasyscyphae) of *Peziza*. Fuckel (1869) raised the Friesian tribe to the status of a genus, as *Dasyscypha*. Unfortunately, Fuckel did not define a characteristic type of paraphysis for the genus, or state which was the type species. *D. calycina sensu* Fuckel has filamentous paraphyses, while the other six species named, including the first, *D. bicolor* (Bull. ex Fr.) Fuckel have lanceolate paraphyses. *Lachnum* Karsten (1871) is based on characters similar to those of *Dasyscypha*, but was limited to include only species with lanceolate paraphyses. Species with filamentous paraphyses, including *D. calycina*, were removed to *Helotium* Fr. *Lachnum* Karst. included *L. bicolor* (Bull. ex Fr.) Karst. and twenty-six other species. Wettstein (1887) followed Karsten.

Boudier (1885) placed stalked species with lanceolate paraphyses in *Dasyscypha*, ignoring *Lachnum*. He created the genus *Trichoscypha* for species with filamentous paraphyses, with a single species, *T. calycina* (Schum. ex Fr.) Boud. Rehm (1887–96) accepted *Lachnum* for species with lanceolate paraphyses, placing related species with filiform paraphyses in *Dasyscypha*. Nannfeldt (1932) pointed out that *Trichoscypha* Boud. was antedated by *Trichoscypha* Hooker and renamed Boudier's genus *Trichoscyphella*. He revived *Lachnum* Karst. for species with an exciple of 'textura prismatica' and usually lanceolate paraphyses. Hahn & Ayers (1934) followed Rehm and named *Dasyscypha calycina sensu* Fuckel as the lectotype for *Dasyscypha*. Dennis (1949), however, followed Nannfeldt in accepting *Trichoscyphella*, but used *Dasyscypha* in place of Nannfeldt's *Lachnum*, since *Dasyscypha* antedates *Lachnum* by two years, and lanceolate paraphyses are not a strictly diagnostic character of *Lachnum* as employed by Nannfeldt. Seaver (1951), following Phillips (1887), placed the hairy inoperculate Discomycetes of conifers in *Lachnella*. His genus *Lachnella* includes, besides this species, others placed in *Dasyscypha* by Dennis. *Lachnella* Fr., however, cannot be used as a generic name for Discomycetes since, as pointed out by Nannfeldt (1932, p. 265) the type species, *Peziza alboviolascens* Alb. & Schw. is a *Cyphella*, and *Lachnella* must be regarded as a Basidiomycete genus. *Trichoscyphella* is accepted here as the appropriate genus for the hairy inoperculate Discomycetes of conifers. The genus as delimited by Nannfeldt appears to form a natural group of rather closely related species: it may be necessary to amend it to include certain little known species with septate ascospores.

There are probably at least seven species of *Trichoscyphella* in Britain, of which three are species with relatively large spores (12μ or more long) occurring mainly on larch, namely (*a*) *T. willkommii* (Hartig.) Nannf., (*b*) the species commonly called *Dasyscypha calycina* (Schum. ex Fr.) Fuckel, and (*c*) *Peziza laricis* Rehm. This last species is little known, and some authors, such as Hiley (1919) and Latour (1950) have maintained that the other two are merely growth forms of a single species. It seems clear, however, from the work of Hahn & Ayers (1934) that they are distinct. Hahn & Ayers gave good descriptions of *Trichoscyphella willkommii* (as *Dasyscypha willkommii*) and of the large-spored saprophyte (as *D. calycina* (Schum. [ex Fr.]) Fuckel), but, as Dennis (1949) pointed out, the latter binomial is invalid for the large-spored larch saprophyte. It seems quite clear, as shown by Wettstein (1887) that Schumacher's fungus is the small-spored species on pine often called *Peziza subtillissima* Cooke. Furthermore, Karsten (1861) definitely used the name *P. calycina* for this small-spored species, so that Fuckel's use of the specific epithet *calycina* for the large-spored larch saprophyte in 1869 cannot be accepted. Seaver (1951) recognized this and, regarding the fungus as a *Lachnella* proposed the new binomial *L. hahniana* Seaver for the fungus described by Fuckel. Since the genus *Lachnella* is unacceptable, *L. hahniana* is redescribed below as a *Trichoscyphella* and a description of *T. willkommii* is added for the sake of comparison. The descriptions (based in part on those of Hahn & Ayers, 1934) are made as a result of an examination of fresh material from various parts of England and Wales and of the collections of these species in the Herbarium of the Royal Botanic Gardens, Kew. Certain important collections from other herbaria were also studied.

Trichoscyphella hahniana (Seaver) Manners, comb.nov.

? *Peziza calycina* γ *laricis* Fr., *Elench. fung.* II, p. 8, 1828.
Lachnella hahniana Seaver, *N. Amer. Cup Fungi*, II, p. 245, 1951.
Dasyscypha calycina auct. non (Schum. [ex Fr.]) Fuck., *Symb. myc.* p. 305, 1869.
Lachnea calycina auct. non (Schum. [ex Fr.]) Gill., *Champ. de France*, p. 71, 1879.
Lachnella calycina auct. non (Schum. [ex Fr.]) Phillips, *Brit. Discom.* p. 241, 1887.
Not *Peziza calycina* Schum. ex Fr.

Apothecia scattered or grouped, erumpent, short stalked, at first globular, closed, opening in a rounded form and expanding under humid conditions to a more or less flat disk with a relatively thin chalky-white rim. Exciple white, of textura intricata, densely covered with hairs. Hairs persistent, cylindrical, thin-walled, minutely granulated, with rounded, slightly swollen tips, 3–4 μ broad. Disk ochraceous-salmon to salmon-orange, 0·5–3 mm. diameter. *Asci* clavate, sometimes slightly swollen towards the obtusely rounded apices, $90–156 \times 9–12\,\mu$, mostly $105–130 \times 9{\cdot}5–11\,\mu$. *Ascospores* 8, obliquely uniseriate, smooth, hyaline, continuous, elongate-elliptic or elliptic-oblong, occasionally fusiform or pointed at one end, $12–21 \times 4{\cdot}5–7{\cdot}5\,\mu$, mostly $13{\cdot}5–19{\cdot}5 \times 5–6\,\mu$. *Paraphyses* exceeding the asci, flexuous-filiform, septate, unswollen at the tips, 1–3 μ broad, intermixed, sometimes very sparsely, with septate filaments 1–4 μ broad, irregularly sub-moniliform towards their tips, which are obtusely rounded or attenuate and sub-acute.

Microconidia $2–5 \times 1–2\,\mu$, continuous, hyaline, elliptic oblong to allantoid, borne on short, subulate, simple or branched sporophores contained in simple or labyrinthiform loculi in erumpent fleshy to waxy whitish stromata. (See Text-fig. 1.)

Saprophytic on dead twigs and bark of *Larix* and occasionally other conifers.

Lectotype: Fuckel, *Fung. Rhen.* 1206 (Herbier Boissier, Botanic Garden, Geneva).

Through the courtesy of the Director of the Geneva Botanic Garden, a portion of the type specimen was examined. As reported by Hahn & Ayers (1934), it was in good condition, and is illustrated in Text-fig. 2. A number of collections of fresh material from the New Forest and Wales were studied, and the measurements quoted above are based on the examination of this material. Eighty-five asci and 680 ascospores were measured in lactophenol, all spores being measured inside the mature ascus. The following collections in the Herbarium of the Royal Botanic Gardens, Kew, proved to be *T. hahniana*.

On *Larix europea*: Ayres, *Myc. Britt.* 56, 1845, as *Peziza calycina* var. *laricis*; Berkeley, *Brit. Fung.* 261, 1836–43, as *P. calycina*; Berkeley, Milton, as *P. calycina* var. *laricis*; Berkeley, Fineshade, Berks, as *P. calycina*; Cooke, *Fung. Brit.* II, 369 (A) Perth, as *P. calycina* (Text-fig. 3); Cooke, Forden (coll. Rev. Vize), as *P. calycina*; Cooke, Ken Wood, as *P. calycina*; Phillips, *Elv. Brit.* 22, as *P. calycina.*

On *Larix leptolepis*: Massee (no details), as *Dasyscypha calycina.*

On *Pinus*: Crossland, Masham 1903, as *D. calycina.*

Cooke, *Fung. Brit.* II, 370 (Text-fig. 4) in the Kew Herbarium has asci 120–135μ long and ascospores 15μ long, mostly with obtuse ends, but has no sub-moniliform paraphyses, and the stalks of the apothecia are longer than is usual in *Trichoscyphella hahniana.* It may be the same fungus as the Berlin specimen of Fuckel, *Fung. Rhen.* 1206, mentioned by Hahn & Ayers (1934). Cooke, *Fung. Brit.* II, 369 (B) Dunkeld (Text-fig. 5) and Crossland, Hebden Bridge, 1893, from Kew lack the long stalks to the apothecia but have no sub-moniliform paraphyses. All these collections probably represent one or more species, or at least varieties, distinct from *T. hahniana*, but it seems inadvisable to describe them as such until fresh material is found. *Peziza laricis* Rehm (1876) was separated from *P. calycina* on the grounds that the asci stained blue with iodine (an unreliable character, see Cooke, 1876). Rehm cited *P. calycina γ laricis* Cooke as a synonym, and Cooke, *Fung. Brit.* II, 370 is probably to be regarded as the type specimen. Unfortunately this exsiccatum is not now in Rehm's herbarium at Stockholm, but if the Kew specimens are regarded as type material, then *P. laricis* Rehm is a valid binomial for the *Trichoscyphella* resembling *T. hahniana*, but differing in the absence of sub-moniliform paraphyses. Seaver (1951) gives *Peziza laricis* as a synonym of *Lachnella willkommii*, but does not state his reasons for so doing. *Dasyscypha willkommii* var. *fuckelii* Bresadola ex Rehm (1887–96) is probably *Trichoscyphella hahniana.* There is a specimen in Rehm's herbarium labelled '*Dasyscypha Fuckelii* Bres. n.sp. = *Dasyscypha calycina* Fuckel nec Schum., ad ramos laricis, Sudtyrol 1883, Bresadola'. This is presumably the type specimen of *D. willkommii* var. *fuckelii*: the binomial *D. fuckelii* does not appear to have been published. The collection resembles *Trichoscyphella hahniana* superficially, but was in very poor condition when examined and no morphological details could be observed.

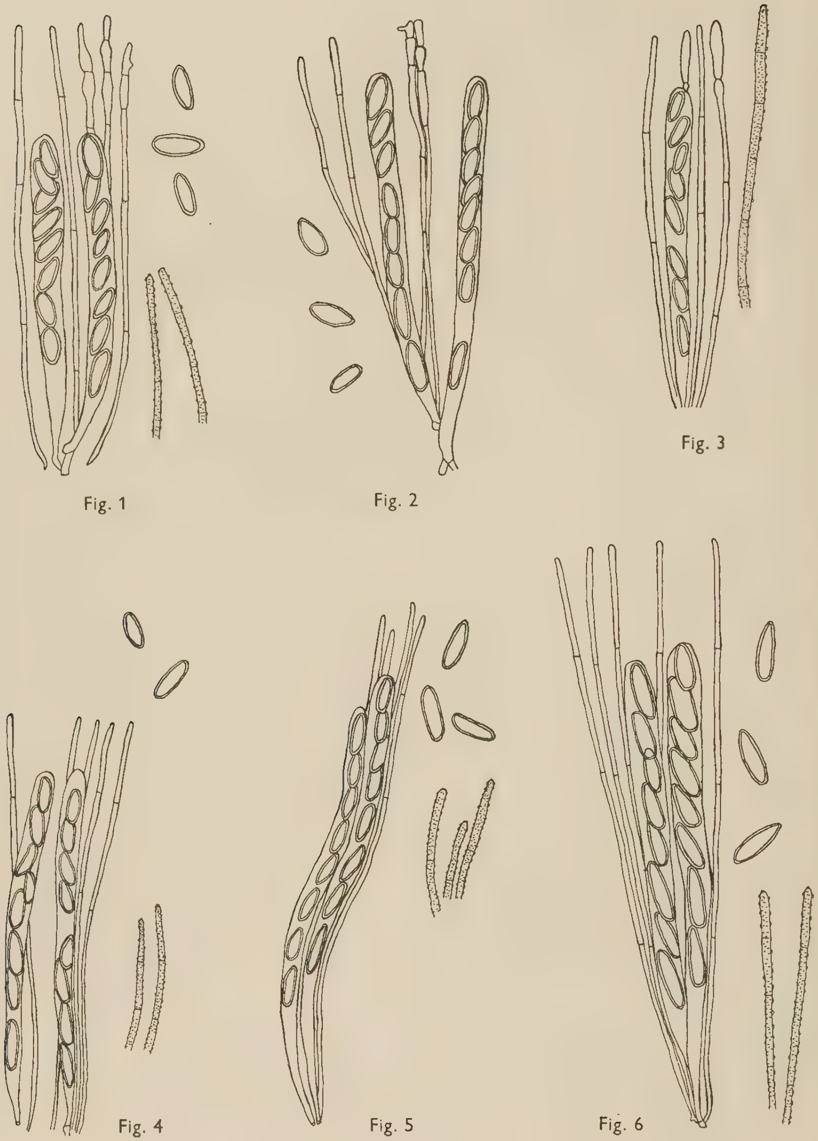


Fig. 1 Fig. 2 Fig. 3 Fig. 4 Fig. 5 Fig. 6

Text-figs. 1–6. Asci, ascospores, paraphyses and excipular hairs of *Trichoscyphella*. Text-fig. 1. *T. hahniana*, from Japanese larch, New Forest, 1950. Text-fig. 2. *T. hahniana* from Fuckel, *Fung. Rhen.* 1206, Herb. Fuckel, Geneva. Text-fig. 3. *T. hahniana*, from Cooke, *Fung. Brit.* II, 369 (A), Herb. Kew. Text-fig. 4. *Trichoscyphella* sp. from Cooke, *Fung. Brit.* II, 370, Herb. Kew. Text-fig. 5. *Trichoscyphella* sp. from Cooke, *Fung. Brit.* II, 369 (B), Herb. Kew. Text-fig. 6. *T. willkommii* from European larch, Coed-y-Brennin Forest, Wales, 1950. All × 440.

Trichoscyphella willkommii (Hartig.) Nannfeldt in *Nova Act. Soc. Sci. Upsal.* Sér IV, **8**, p. 299, 1932.

Peziza willkommii Hartig, *Wichtig Krankh. Waldbäume*, p. 98, 1874.

Dasyscypha willkommii (Hart.) Rehm, 26*ste Ber. Naturh. Ver. Augsberg*, p. 19, 1881.

Helotium willkommii (Hart.) Wettstein, *Bot. Zbl.* **31**, 285, 1881.

Trichoscypha willkommii (Hart.) Boud., *Discom. d'Europe*, p. 125, 1907.

Apothecia scattered or grouped, erumpent, short stalked, at first globular, closed, opening in a rounded form and expanding under humid conditions to a convex disk with a thick, rather irregular chalky-white rim. Exciple white, of textura intricata, densely covered with hairs. Hairs persistent, cylindrical, thin-walled, minutely granulated, with rounded, slightly swollen tips, 3–4 μ broad. Disk apricot-orange or apricot-buff, fading to light buff, 1–4(–6) mm. diameter. *Asci* clavate, sometimes slightly swollen towards the obtusely rounded apices, 90–156 × 9–12 μ, mostly 105–130 × 9·5–11 μ. *Ascospores* 8, obliquely uniseriate, smooth, hyaline, continuous, elongate-elliptic, fusiform, obtuse or pointed at one or both ends, 13·5–25·5 × 4·5–7·5 μ, mostly 15–23 × 5–7 μ. *Paraphyses* exceeding the asci, flexuous-filiform, septate, obtuse, not swollen at the tips, or only very slightly so, 1–3 μ broad (up to 4 μ at the tip).

Microconidia 2–8 × 1–2 μ, continuous, hyaline, elliptic-oblong to allantoid, borne on short, subulate, simple or branched sporophores contained in irregular labyrinthiform loculi in erumpent fleshy to waxy whitish stromata.

Parasitic on *Larix*, occurring only in the immediate vicinity of cankers.

Type: The type material collected by Hartig no longer exists (Hahn & Ayers, 1934).

A number of collections of fresh material from the New Forest and Wales were studied, and the measurements quoted above are based on the examination of this material. Fifty-five asci and 440 spores were measured in lactophenol, all spores being measured inside the mature ascus. The following collections in the Herbarium of the Royal Botanic Gardens, Kew, proved to be *Trichoscyphella willkommii*.

On *Larix europea*: Berkeley, Glamis, 1874, as *Peziza calycina*; Cavarra & Pollacci, *I Fung. par.*, 433, 1912, as *Dasyscypha calycina*; Cooke, Dunkeld, as *P. calycina*; Massee (no details) as *D. calycina*.

All the exsiccati, like those listed under *Trichoscyphella hahniana*, were readily determinable when small portions of apothecia were teased out after boiling in lactophenol. A number of other exsiccati examined could not be determined, owing to their poor stage of preservation. Fresh collections may usually be assigned to their correct species in the field, the thick exciple and pale disk of *T. willkommii* being quite distinctive. These characters are, however, of no value in the examination of old herbarium specimens. In any case, determinations must be confirmed from microscopical characters, of which the length of the ascus and the presence or absence of sub-moniliform paraphyses are the most reliable. Much confusion has been caused in the past because any apothecium associated with cankers has been placed in *T. willkommii*. This species only occurs on more or less active cankers, while *T. hahniana* is found on old cankers as well as on dead twigs (Pl. 23, fig. 2).

There are certain cultural differences between the two species. In both, the ascospores are septate on germination, but the germ tube branches more profusely in *Trichoscyphella willkommii* than in *T. hahniana*. Growth on 3 % malt agar of mycelium derived from single ascospores is slow but steady, with an optimum growth rate at 20° C. (see Table 1). The growth rate was determined by placing 2 mm. square blocks of inoculum from young cultures on 3 % malt agar. Two plates were incubated at each temperature and in each, two diameters at right angles to each other of the resulting colony were measured and the mean recorded. The experiment was repeated with a number of cultures of each species, similar results being obtained in each case.

Table 1. *Growth rate of* Trichoscyphella hahniana *and* T. willkommii

	Average mean diameter in mm. after 16 days					
Inoculum	5° C.	10° C.	15° C.	20° C.	25° C.	30° C.
T. hahniana, culture 18/3	5·5	12·5	18·5	20·5	11	0
T. willkommii, culture 18/1	3·5	12	20·5	24·5	17	0

In culture, *Trichoscyphella hahniana* produces a low, mealy type of aerial mycelium, whereas *T. willkommii*, which usually grows slightly faster, produces a dense white velvety aerial mycelium. In tube culture both species form a plug of mycelium at the top of the tube and in older cultures a pinkish buff colour develops and imperfect fructifications are formed. Cultures of both species were established on green twigs of *Larix europea*, sterilized by autoclaving or by exposure to propylene oxide vapour, as described by Rishbeth (1950) and placed on damp cotton-wool in boiling tubes. Within a few weeks the twigs were covered with white mycelium, which was especially profuse in the tubes inoculated with *Trichoscyphella willkommii*. The tubes were maintained for 9 months in an incubator at 22·5° C., but produced only microconidia. They were then transferred to a well-lit laboratory bench and within a month several tubes inoculated with *T. hahniana* produced small apothecia. These were not observed in tubes inoculated with *T. willkommii*, and this difference between the two species cannot at present be accounted for. (See Pl. 23, fig. 1.)

THE INOCULATION OF TREES IN A FROST-FREE LOCALITY

The results of previous inoculation experiments designed to determine whether *Trichoscyphella willkommii* is the cause of larch canker have been conflicting. Hartig (1880) inoculated young European larches with ascospores through small wounds and obtained local lesions and fructifications. Attempts to infect uninjured trees failed and he concluded that the fungus was a wound parasite. Massee (1902) obtained cankers on European, Siberian and Japanese larches and Scots pine by artificial infection and found that cankers resulted when ascospores were placed on the mucilaginous excretion of *Chermes abietis* in the spring. Massee's inoculum may well have been mixed as *Trichoscyphella willkommii*, *T. hahniana* and the Discomycete usually called *Erinella pommeranica* Ruhland all occur under the name of *Dasyscypha calycina* in Massee's herbarium. Hiley (1919, p. 50) inoculated European larch by placing mycelium from cultures in small knife-slits in the bark of young trees. Using 'the parasitic form of *D. calycina*', i.e. *Trichoscyphella willkommii*, he obtained normal cankers bearing fructifications. Plassmann (1927) obtained some positive, but some negative, results, perhaps due to a failure to distinguish between *T. willkommii* and *T. hahniana* and Gaisberg (1928) obtained negative results, perhaps for the same reason.

Hahn & Ayers (1938) obtained negative results with *T. willkommii* on Douglas fir, but later (1943), using Hiley's inoculation method, they obtained cankers with this species on European and Japanese larches and several American species of larch. Negative results were obtained on these hosts with *T. hahniana* and several other large-spored Trichoscyphellas. They reported that Mr T. R. Peace, working at Oxford, obtained die-back due to girdling cankers in European larch saplings using four cultures of *T. willkommii*, two American and two British, supplied by them. The trees were in cold frames and thus protected from frost.

Methods

The object of the experiments to be described was to determine whether *Trichoscyphella willkommii* or any related species could cause canker or die-back on European larch grown out of doors under British conditions in a locality where the possibility of frost damage was excluded. Mr T. R. Peace (Forestry Commission, Research Branch) kindly made available for inoculation purposes a number of young trees of European larch which he had planted at Red Lodge, Trewidden Estate, near Penzance, Cornwall, on a site far from any other larch. In this locality frost is very rare and the chances of frost damage are therefore negligible. During the course of the experiments there were never more than two or three frosty nights per year and no frost sufficient to cause any damage to larch.

The method of inoculation was similar to that employed by Hahn & Ayers (1938). The spot to be inoculated was rubbed with cotton-wool soaked in methylated spirit to remove debris and surface-sterilize the bark. A longitudinal slit 2 cm. long was then made in the bark with a sterile scalpel, the bark on one side of the slit was raised with the tip of the scalpel and the inoculum was inserted by means of a second, small scalpel. The cut edges were then pressed down over the inoculum. The wound was covered with sterile cotton-wool soaked in sterile water and held in place by a 2 in. band of 'Cellophane' or greaseproof paper bound above and below the wound with string. Control trees were inoculated with sterile 3 % malt agar, the experimental trees with cultures growing on the same medium.

Each culture used for inoculation was derived from a single ascospore, a modification of the single spore isolation method described by LaRue (1920) being employed. A Petri dish containing 3 % malt agar (made up with 5 % agar to give extra stiffness) was inverted and an apothecium placed in the lid. The apothecium was raised on slides so that the disk was about 2 mm. from the surface of the agar. The dish was then incubated overnight at 22·5° C. During this time the ascospores were discharged on to the agar and, if the distance between the apothecium and the agar was correct, each spore was well separated from its neighbours. The apothecia were then removed and the dishes incubated for a further 24 hr. to allow the spores to germinate. Disks of agar containing single germinated spores were then cut out with a dummy objective similar to that described by LaRue (1920) with a cutter having a diameter equal to that of the $\frac{2}{3}$ in. field. Each disk was then transferred on a flattened platinum needle to a separate culture tube. The operation was performed in an inoculation chamber and the cutter was sterilized by flaming before making a cut. The amount of contamination was negligible and cultures were established from over 90 % of the spores isolated.

Results

The results were assessed according to the following scale:

0 Wound healing normally.
1 Wound healing slowly, sometimes with slight necrosis.
2 Wound not healing, bark slightly sunk around wound, young apothecia present.
3 As 2, but in addition, the stem or branch beyond the point of inoculation is dead or dying.

Plus and minus signs were sometimes used for intermediate states. Forty-eight trees were inoculated on 28 August 1950; of these, half, which were of Scottish origin (Glenlivet, Banffshire), were more vigorous than the others raised at Wareham Nursery (Dorset) which were of Swiss alpine origin. These trees had previously been inoculated on their main stems by Mr Peace, with apparently negative results, so an inoculation was made on each tree towards the base of a vigorous lateral 2–3 years old. The inoculum consisted of cultures made from material collected in the New Forest in 1950. *Erinella pommeranica* was included because this species (which is related to *Trichoscyphella* and may belong to that genus (Dennis, 1949)) is often associated with cankers on Scots pine in the New Forest, and is at present under investigation. The trees were examined on 4 January 1951, when the wrappings over the wounds were removed. No disease symptoms were visible on this date. The results of two further examinations on 18 April 1951 and 5 September 1951 are summarized in Table 2. The results recorded on the two dates were almost identical.

On 5 September 1951 all these trees were removed except for those marked with an asterisk in Table 2, which were examined on 10 January 1952, 2 April 1952 and 2 September 1952, with the results shown in Table 3. The results of this experiment indicated that *T. willkommii*, but not *T. hahniana* or *Erinella pommeranica*, was capable of causing canker and die-back on European larch under the conditions of the experiment.

Further inoculations were carried out on 18 April 1951, to confirm these results and determine whether alpine strains of larch were more liable to infection than Scottish strains. Forty-eight trees (all European larch) were inoculated: twenty-four had been planted as 1 + 1 transplants on 27 March 1950 and were Scottish grown, of Loch Ard origin (Forestry Commission, ident. no. 46/91), while twenty-four had been planted as 1 + 1 transplants on 17 February 1950 and were English grown, of alpine origin (Forestry Commission, ident. no. 47/53). These trees had not been inoculated previously, so were inoculated on the main stem on wood 1–2 years old. The inoculum was obtained from various sources. Cultures 15/5, 103/3, 103/5 and 203 were made from material collected in the New Forest in 1950. Cultures 17/1 and 18/1 from Coed-y-Brennin Forest, Wales, and cultures 20/1 and 20/2 from Radnor Forest, Wales, were made from material collected on a tour of certain Welsh forests made in September 1950. Cultures 24 and 25, kindly supplied by Mr Peace, were obtained by him from the Centralbureau voor Schimmelcultures, Baarn, Holland. These last two cultures produced low, smooth colonies on 3 % malt agar, unlike those produced by freshly isolated cultures of either *Trichoscyphella willkommii* or *T. hahniana*.

Table 2. *Inoculations at Penzance in 1950*

Culture	Source	No. of trees inoculated	Results			
			Scottish		English	
Trichoscyphella willkommii						
13	European larch	4	2*	0	2	0
15/5	European larch	4	2*	2*	1	3
103/3	Japanese larch	4	3	3	3	3
103/7	Japanese larch	4	3	3	3*	3
T. hahniana						
16	European larch	8	0 0	0 0	0 0	0 0
103/5	Japanese larch	4	0	1	0	1
103/8	Japanese larch	4	0*	0	0	0
Erinella pommeranica						
201/1	Scots pine	8	0 0	0 1	0 1	1 1
Control: sterile agar	—	8	0* 0	0 0	0 0	0 0

* These trees were retained for further observation.

Table 3. *Inoculations at Penzance in 1950 (further observations)*

Culture	Source	Date of observation		
		10. i. 52	2. iv. 52	2. ix. 52
Trichoscyphella willkommii				
13	European larch	2	2	2+
15/5	European larch	2 1	2 1	2+ 0
103/3	Japanese larch	2	2	3*
103/7	Japanese larch	2	3	3
T. hahniana				
103/8	Japanese larch	0	0	0
Control: sterile agar	—	0	0	0

* This tree is shown in Pl. 23, fig. 5.

Infection developed rather slowly. None was apparent on 5 September 1951 and only a few trees showed signs of infection on 10 January 1952. Symptoms developed more rapidly from then on, and the results obtained on 2 April 1952 and 2 September 1952, which did not differ much from each other, are summarized in Table 4.

These results confirm those obtained in the earlier experiment. Culture 18/1, from European larch was only slightly pathogenic, while culture 25 had completely lost its pathogenicity. The two trees infected by *T. hahniana* were both inoculated with culture 103/5, which is in certain respects intermediate between the two species (an infection grade of 1 is not regarded as constituting certain evidence for infection). The figures suggest that some of the cultures from the New Forest, established more than a year before inoculation, may have lost some of their pathogenicity, but further work is required to determine this point.

Table 4. *Inoculations at Penzance in* 1951

Culture	Source	No. of trees inoculated	Results			
			Scottish		Alpine	
Trichoscyphella willkommii						
15/5	European larch	4	1	0	2	2
18/1	European larch	4	0	1	1	0
20/1	European larch	4	2	0	2	3—*
20/2	European larch	4	2	2	2	2
103/3	Japanese larch	4	1	0	3*	2
25	European larch	4	0	0	0	0
T. hahniana						
17/1	European larch	4	0	0	0	0
103/5	Japanese larch	4	2	2	0	0
24	European larch	4	1	1	0	0
Erinella pommeranica						
203	Scots pine	4	0	0	0	0
Control: sterile agar	—	8	00	00	00	00

* These trees are shown in Pl. 23, figs. 3 and 4.

These experiments indicate that *Trichoscyphella willkommii*, but not *T. hahniana*, can cause cankers on European larch in the absence of frost and that Scottish strains may be rather less susceptible to the fungus than alpine strains. Of thirty-two trees inoculated with *T. willkommii*, twenty-four became infected; of twenty-four trees inoculated with *T. hahniana*, two became infected (both were trees inoculated with culture 103/5, see above). Trees inoculated with cultures 24 and 25 are not included in these totals. There is no evidence that *Erinella pommeranica* can parasitize larch, but inoculum from Japanese larch is at least as virulent on European larch as inoculum from the latter species.

Discussion

The findings reported in this paper confirm those of Hahn & Ayers (1934, 1943) in most respects. It has been suggested that *Trichoscyphella hahniana* and *T. willkommii* appear distinct in the United States merely because intermediate forms have not yet become introduced there. However, British collections can almost invariably be assigned without difficulty to one species or the other, even when the collections are herbarium specimens of considerable age. Hahn & Ayers' nomenclature cannot, however, be accepted, for the reasons already given, and it is hoped that the binomials advocated in this paper will come into general use.

The inoculation experiments have shown that *T. willkommii*, but not *T. hahniana*, can cause cankers in the absence of frost, but it is not yet clear for how long such cankers may persist and grow in the absence of frost, and it is hoped to obtain data on this point in due course. The conditions of inoculation employed were artificial: there is no evidence that cankers are normally produced around mechanically caused wounds. It seems more probable, in view of the correlation between frost damage and canker noted by Day (1931) and Latour (1950), that in nature, wounds caused by frost form the normal channel of entry, and this possibility is now being investigated.

The conditions required for the persistence and growth of a canker, once formed, are also in need of further investigation. Hahn & Ayers (1943) working in a frost-free area obtained only eleven cankers from 212 infections, and it may well be that both the presence of *T. willkommii* and periodical exposures to low temperatures are required, as suggested by Langner (1936) and Münch (1936). Day & Peace (personal communication) only obtained cankers in a very small proportion of the larches artificially frost damaged by them, perhaps because of the small size of the trees employed.

I wish to express my thanks to the Directors of the Royal Botanic Gardens, Kew, the Conservatoire, Geneva and the Naturhistoriska Riksmuseet, Stockholm for the loan of herbarium specimens, to Dr R. W. G. Dennis for advice on taxonomic problems, to Mr T. R. Peace and other Forestry Commission officers for their kindness in supplying material and information, and to Mr F. A. Barrett for photography. My thanks are also due to the Agent for the Trewidden Estate, Penzance, for providing facilities at Red Lodge Nursery. This work was carried out in part with the aid of a grant from the Forestry Commission, to whom acknowledgement is made.

REFERENCES

Boudier, E. (1885). Nouvelle classification naturelle des Discomycètes charnus, connus généralement sous le nom des Pezizes. *Bull. Soc. mycol. Fr.* **1**, 91–120.

Cooke, M. C. (1876). Observations on *Peziza calycina*. *Grevillea*, **4**, 169–172.

Day, W. R. (1931). The relationship between frost damage and larch canker. *Forestry*, **5**, 41–56.

Day, W. R. (1937). The dying of larch: a note on Professor E. Münch's monograph *Das Lärchensterben*. *Forestry*, **11**, 109–116.

Day, W. R. & Peace, T. R. (1934). The experimental production and the diagnosis of frost injury of forest trees. *Oxf. For. Mem.* **16**, 60 pp.

Dennis, R. W. G. (1949). A revision of the British Hyaloscyphaceae with notes on related European species. *Mycol. Pap.* **32**, 97 pp.

Fries, E. (1822). *Systema Mycologicum*, **2**, 89.

Fuckel, L. (1869). *Dasyscypha* Fuckel. *Jb. nassau. Ver. Naturk.* **23–24**, 305.

Gaisberg, E. von (1928). Beiträge zur Biologie des Lärchenkrebspilzes, *Dasyscypha willkommii* Hartig. *Mitt. wurttemb. forstl. VersAnst.* 24 pp.

Hahn, G. G. & Ayers, T. T. (1934). Dasyscyphae on conifers in North America. I. The large-spored, white excipled species. *Mycologia*, **26**, 73–100.

Hahn, G. G. & Ayers, T. T. (1938). Failure of *Dasyscypha willkommii* and related large-spore species to parasitize Douglas Fir. *Phytopathology*, **28**, 50–57.

Hahn, G. G. & Ayers, T. T. (1943). Role of *Dasyscypha willkommii* and related fungi in the production of canker and die-back of larches. *J. For.* **41**, 483–495.

Hartig, R. (1880). Die Lärchenkrankheiten insbesondere der Lärchenkrebspilz. *Untersuch. forstb. Inst. München.* **1**, 63–87.

Hiley, W. E. (1919). *The fungal diseases of the common larch.* 204 pp. Cambridge.

Karsten, P. A. (1861). *Synopsis Pezizarum et Ascobolorum Fenniae.*

Karsten, P. A. (1871). *Mycologia fennica, Pars prima, Discomycetes.*

Langner, W. (1936). Untersuchungen über Lärchen-, Apfel- und Buchen-krebs. *Phytopath. Z.* **9**, 111–145.

LaRue, C. D. (1920). Isolating single spores. *Bot. Gaz.* **70**, 319–320.

Latour, J. M. (1950). Intervention de la gelée dans la formation du chancre du mélèze d'Europe (*Larix decidua* Miel.). *Bull. Soc. for. Belg.* **57**, 239–241.

Massee, G. (1902). Larch and spruce fir canker. *J. Board Agric.* **9**, 176.

Münch, E. (1936). Das Lärchensterben. *Forstwiss. Zbl.* **58**, 469–494, 537–562, 581–590, 641–671.

Nannfeldt, J. A. (1932). Studien über die Morphologie und Systematik der nicht-lichenisierten inoperculaten Discomyceten. *Nova Acta Soc. Sci. upsal.* Ser. 4, **8**, 1–368.

Phillips, W. (1887). *A manual of the British Discomycetes.* 462 pp. London.

Plassmann, E. von (1927). *Untersuchungen über den Lärchenkrebs.* 88 pp.

Rehm, H. (1876). Notes on *Peziza calycina* Schum. *Grevillea*, **4**, 169.

Rehm, H. (1887–96). *Rabenhorst's Kryptogamenflora*, Bd. 1, Abt. III, *Ascomyceten.*

Rishbeth, J. (1950). Observations on the biology of *Fomes annosus* with particular reference to East Anglian pine plantations. I. The outbreaks of diseases and ecological status of the fungus. *Ann. Bot., Lond.*, N.S., **14**, 365–383.

Seaver, F. J. (1951). *The North American Cup-fungi* (*Inoperculates*). 428 pp.

Wettstein, R. von (1887). Ueber *Helotium Willkommii* (Hart.) und einige ihm nahe stehenden Helotium-Arten. *Bot. Zbl.* **31**, 285–287, 317–321.

EXPLANATION OF PLATE 23

Fig. 1. Nine-month cultures of (A) *Trichoscyphella willkommii* and (B) *T. hahniana* on sterilized larch twigs.

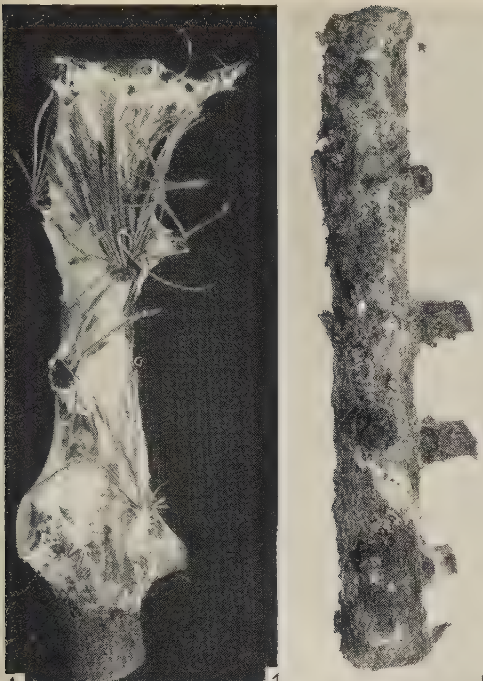
Fig. 2. Apothecia of (A) *T. willkommii* on canker on European larch and (B) *T. hahniana* on an adjacent dead lateral branch.

Fig. 3. Canker on European larch at Penzance, 2½ years after inoculation with *T. willkommii.*

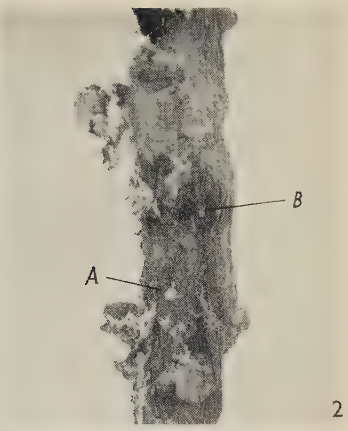
Fig. 4. Die-back of the leader in European larch at Penzance. The tree, photographed after needle fall, had died back to the point of inoculation (A).

Fig. 5. Die-back in European larch at Penzance, 3 years after inoculation with *T. willkommii* at the base of a lateral branch.

(*Accepted for publication* 13 *December* 1952)

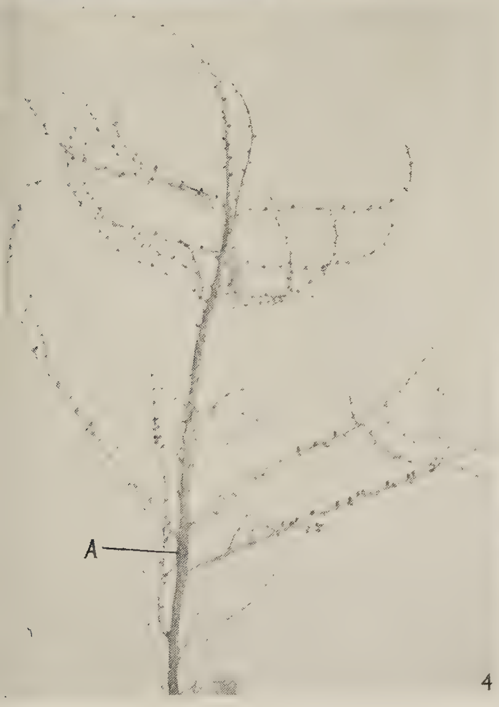


1



2

3



4

5

CHANGES IN ATMOSPHERIC SPORE CONTENT: DIURNAL PERIODICITY AND THE EFFECTS OF WEATHER

By J. M. HIRST

Rothamsted Experimental Station, Harpenden, Herts

(With Plate 24 and 5 Text-figures)

The use of a power-operated suction trap, in which spores are impacted on the sticky surface of a slowly moving slide, made it possible to estimate accurately the concentration of different kinds of spore in the air at any given time. In an open field at 2 m. above ground the catches often consisted predominantly of small hyaline spores, of types rarely caught by other kinds of trap. Changes in the types of spore caught at different times of day showed that some species have well-defined diurnal periodicity; the catches also reflect changes in weather.

In dry weather, pollen grains and spores of such fungi as *Cladosporium*, *Erysiphe*, *Alternaria*, smuts and rusts, are the main components of the air spora; they are most abundant in the afternoon and least in the early morning. *Phytophthora infestans* and *Polythrincium trifolii* also occur, but reach their maximum concentration before noon. Prolonged rain removes most of these spores, and for a time there are few in the air. Within a few hours of the start of rain, the typical dry-air spora is replaced by hyaline spores; few of these can be identified, but they include splash-dispersed types, ascospores and basidiospores. Except after rain this damp-air spora occurs in quantity only at night when dew is formed, and the greatest concentration is reached between midnight and dawn.

Basidiospores were at times the dominant type of spore. Hyaline forms, often mainly *Sporobolomyces* sp., rapidly reached large numbers in the early hours of the morning, particularly when there was heavy dew, and then disappeared almost completely. Coloured basidiospores mainly from the Agaricales were also commonest at night, but their occurrence depended less on weather than the hyaline forms and their diurnal periodicity was less pronounced.

INTRODUCTION

Knowledge of the time and weather associated with spore dispersal is often required to understand the etiology and epidemiology of plant diseases and may assist in formulating forecasting and control methods. Gregory (1951, 1952*a*) and Gregory & Stedman (1953) have shown that many of the current trapping methods allow neither a true quantitative nor qualitative assessment of the air-borne spore flora or 'air spora'. With a few exceptions, for instance Keitt & Jones (1926), plant pathologists have used sticky microscope slides freely exposed in various attitudes relative to the wind. In routine trapping, slides have usually been exposed for 24 hr., although shorter periods have been used for the study of particular organisms.

Allergists are concerned with the changes of concentration of plant spores in the air and their effect on the severity of attacks of rhinitis and asthma. Hay-fever may be related to characteristic times of pollen liberation (Hyde & Williams, 1945, 1946; Jones, 1952). There have been few investigations of short-period fluctuations in the concentration of fungal allergens in the air, but, in Wisconsin, Heise & Heise (1950) found the spore load of pollens and *Alternaria* varied with time of day, locality and altitude. They explained these variations as due to local air movements on the western shores of Lake Michigan. Alvarez & Castro (1952) exposed Petri dishes for 15 min. at hourly intervals, first in the open air, and later with the dish at the bottom of a tall box which could be shut to allow the spores in a known volume of air to sediment on to a nutrient medium. The two methods gave widely different results. The differences might occur because the efficiency of trapping by the Petri dish changed at different wind speeds, or, as the two tests were not simultaneous, because the populations sampled differed. The only discernible effect of weather on the catches of a volumetric sampler described by Hawes, Small & Miller (1942) was a reduced catch after rain, at other times the counts showed a general downward trend throughout a 24 hr. run. This probably occurred because the very narrow orifice became blocked by large particles which altered the suction rate and the penetration of spores. Most routine aerobiological surveys by allergists have relied on either the exposure for 24 hr. periods of a 'gravity slide'; or the exposure for a much shorter period, usually about 15 min., of a Petri dish containing nutrient medium. In the latter method the resultant colony count has been assumed to represent the spore concentration for the day. Interpreting the catches on such traps is difficult because their efficiency depends, not only on the position and dimensions of the trap, but on the size and shape of the spores, and on the wind speed at the time of deposition. The freely exposed surface traps are highly selective and favour the heavier spores; in the Petri dish method selection is increased by the limited number of species which will grow on the medium, by antagonism between colonies and by the overgrowth of other colonies by some quick-growing moulds. Such methods fail to show any changes that may occur over short periods and, as Durham (1944) has recognized, may give a false interpretation of the daily mean concentration.

METHODS

This paper deals with preliminary results obtained by operating continuously an automatic volumetric spore trap (Hirst, 1952) at a height of 2 m. above ground level at Rothamsted Experimental Station, Harpenden during June to September in 1951 and 1952. This trap was designed to improve and simplify the previous methods for making successive 2-hourly estimates of the concentration in the air of the larger-spored plant-pathogenic fungi. Air is sucked at 10 l./min. through an orifice 14 × 2 mm., the inner end of which is approximately 0·5 mm. from the sticky surface of the microscope slide on which the spores are impacted. In order to separate the spores caught at different times the slide is moved past the orifice at a constant rate of 2 mm./hr. Spores and other particles caught during a 24 hr. exposure are deposited on a trace measuring approximately 48 × 14 mm. (Pl. 24, fig. 1). The trap was designed to give a high efficiency of collection as well as of impaction or retention. The trap has been calibrated in wind-tunnel tests using spore clouds of known concentration.

Several of the methods have been modified since the automatic volumetric spore trap was first described, others need additional description.

Preparation of slides. Vaseline remains the most satisfactory adhesive for use in various types of weather, despite a troublesome artefact, the presence of circular depressions in the Vaseline film thought to be caused by water droplets. Slides are now prepared for exposure by coating 55 mm. of the length of the slide with a 10 % solution of Solvar* which is allowed to dry. A thin film of Vaseline is then applied to the coated area, melted and smoothed out by the slow movement of a smooth edge. Melting the Vaseline appears to remove stresses which lead to splits in the film after mounting; unfortunately it also causes some deterioration of optical properties. Before exposure the Vaseline is removed from strips 3 mm. wide along the edges of the slide, as these parts do not pass under the orifice.

Mounting slides. Sealing the slides with wax was found to be only partially satisfactory and has been replaced by a water-soluble plastic (Downs, 1943). After exposure, the slide is gently warmed on a hot-plate suitable for spreading wax microtome sections. When cool again it is inverted and lowered on to a 2 × $\frac{7}{8}$ in. cover-glass bearing sufficient of the mountant to spread easily over its whole area. The mountant is a mixture of 50 parts of a 20 % solution of Solvar with 25 parts of lactic acid and 25 parts of a 6 % solution of phenol. After labelling, the mount is left to dry for about 2 days when, without any further sealing, it will have set sufficiently for handling and storage, although with care it may be handled before this. At present no stains are incorporated in the mountant, as natural colour aids spore identification. For examining certain types of spore the use of stains or phase contrast may be an advantage.

Counting spores. With an orifice 2 mm. wide and the slide moving at 2 mm./hr., any straight line across the width of the slide will remain under the orifice for 1 hr. Increase in the width of the traverse when scanning under the microscope reduces the accuracy of estimating the time of deposition of spores. Thus counts on a traverse 1 mm. wide would indicate the average deposit during a period of 2 hr. Usually the number of spores per unit area of slide is much greater than on the freely exposed surface traps. Therefore reliable counts may be made by scanning much smaller areas than usual with the earlier traps. For instance a traverse 20 μ wide has at times contained over a thousand hyaline basidiospores. Further evidence on this point will be presented elsewhere, when different trapping methods in the open air will be compared (Gregory, Hirst & Last, *unpublished*).

---

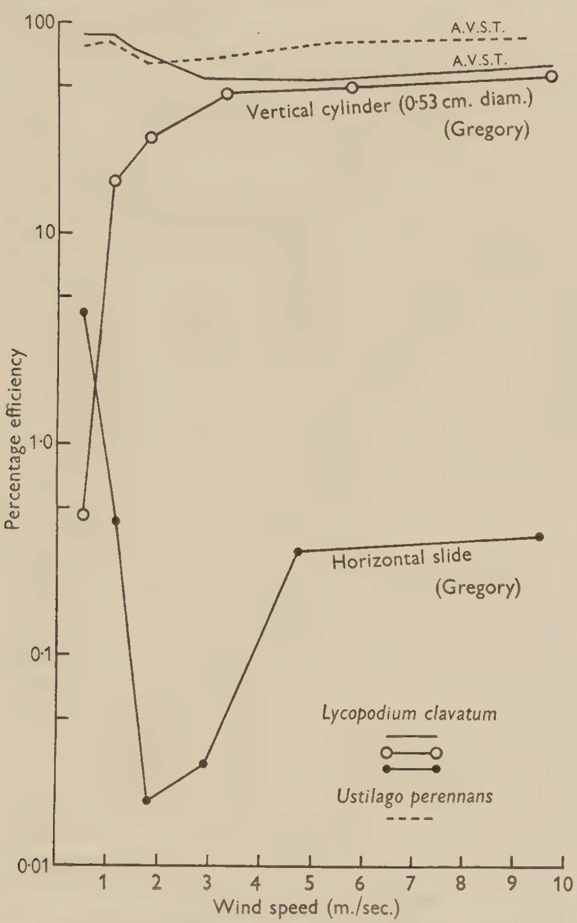
* Solvar is the name given to a range of partially hydrolysed polyvinyl acetate preparations marketed by Shawinigan Ltd., Marlow House, Lloyds Avenue, London, E.C. 3. Solvar 357 has normally been used but Solvar 2105 and 3515 also appear suitable.

The exact procedure of counting adopted depends on the types of spore investigated, their frequency, and the length of the interval at which counts are required. The simultaneous counting of several spore types is facilitated by mounting hand-operated counters close to the stage and focusing controls of the microscope. Low-power objectives are sufficient for some pollens and the larger-spored fungi. However, if there are enough spores, it is usually preferable to count a narrower trace under a high power objective, thus ensuring that spores are not overlooked (particularly when close to 'droplets' or dust particles), or wrongly identified. For minute hyaline spores and basidiospores, scanning under an oil-immersion objective is required.

Daily mean concentrations may be estimated relatively quickly by counting spores on traverses parallel to the direction of slide movement (long traverses), but corrections for the efficiency of the trap can then be applied only on the basis of the daily mean wind speed. For estimates at shorter intervals, or for more accurate daily mean values, counts are made on traverses at right angles to the direction of slide movement (short traverses) at positions representative of known times. Corrections for the efficiency of the trap are applied, where possible, using the mean wind speed during a limited period about these times, and the resultant estimates are averaged if a daily mean value is required. The results in this paper have all been obtained by the short traverse method. Counts have usually (not in Text-fig. 2) been made at the position of the centre of the orifice at each odd hour, and the mean wind speed used has been that between the adjacent even hours. The slide was changed at 0900 daily and as the estimate at this time is therefore liable to be less accurate than the rest, it has been reinforced by counts at 0800 and 1000. All times given are G.M.T.

Trapping efficiency. The trap was designed to study the dispersal of large-spored plant-pathogenic fungi, and it was not expected to retain completely all recognizable spores entering the trap, as does the Cascade Impactor (May, 1945; Gregory 1952*a*). Simultaneous operation of these two traps in the open, however, has shown that the Automatic Volumetric Spore Trap retains small spores with a considerably higher efficiency than was expected from the known performance, under similar circumstances, of the second orifice of the Cascade Impactor which is of similar cross-section. This is attributed to the fact that both the long sides of the orifice extend to within the same distance from the slide, whereas in the Cascade Impactor the first two jets are made with one side shorter than the other to deflect the air stream towards the next jet. The deflexion reduces the velocity of the air stream close to the sticky surface and fewer spores are impacted at these stages.

Trapping efficiency in the open air varies with the wind speed, because the suction rate is not altered with the wind speed to maintain isokinetic sampling. Calibration tests have therefore been made in turbulent air in the wind tunnel over the range of wind velocity from 0·5 to 10·0 m./sec. Two test particles have been used, spores of *Lycopodium clavatum* and of *Ustilago avenae*, 32 and 6·5 μ in diameter, respectively. Text-fig. 1 shows the results of these tests, together with the efficiencies found by Gregory for deposition on gravity slides and vertical cylinders 0·53 cm. in diameter. At most wind speeds the suction trap catches *Ustilago* more efficiently than *Lycopodium*; this is probably explained by the more efficient collection of the smaller than those of the larger particles at the orifice where sampling is not isokinetic. In the subsequent text or figures, [L] or [U] following



Text-fig. 1. Comparative efficiency of traps in turbulent wind tunnel, using *Lycopodium clavatum* (32 μ diam.), or *Ustilago avenae* (6·5 μ diam.) as test particles, and wind speeds between 0·5 and 10·0 m./sec.

the name of a spore group denotes that corrections have been applied based on the calibration tests and the mean wind speed at the time. Some spores cannot be referred to either test particle, because of peculiar shape, variable size, or clumping. These uncorrected estimates have been joined on all diagrams by broken lines. As the trap is never 100% efficient the catch always underestimates the actual number present.

Expression of results. In expressing results it is at present difficult to satisfy both the allergist interested in the number of spores entering the respiratory system and the plant pathologist requiring the number of spores deposited on the surface of susceptible hosts. The deposition of spores on the leaves and stems of a crop is not yet understood, although the principles which Gregory has found to operate on artificial surfaces will probably be found to apply. At present it is best to express results in terms of the number of spores per cubic metre of air, although the 'area dose' (Landahl & Hermann, 1949) may well become a more convenient measure when more is known of the deposition of spores on crops.

The number of spores counted on a traverse of known area can readily be converted into an estimated number per cubic metre from the known suction rate (0·6 $m.^3$/hr.), the cross-sectional dimensions of the orifice (2 × 14 mm.) and the rate of movement of the slide (2 mm./hr.).

Identification of spores. The use of suction traps greatly increases both the total catch of spores and the range of types caught, the increase is greatest with the small and hyaline types, few of which had previously been seen in trap catches. Many of these unfamiliar spores are difficult to identify. Some of the highly characteristic spores such as those of *Phytophthora infestans*, *Venturia inaequalis*, *Polythrincium trifolii*, some uredospores, and the basidiospores of some agarics can be identified specifically. For most spores a generic or an even wider grouping is the best which can be achieved at present. These studies aimed at determining the extent and frequency of changes in spore load of a wide variety of spore types and no effort was made to press identification to its limits.

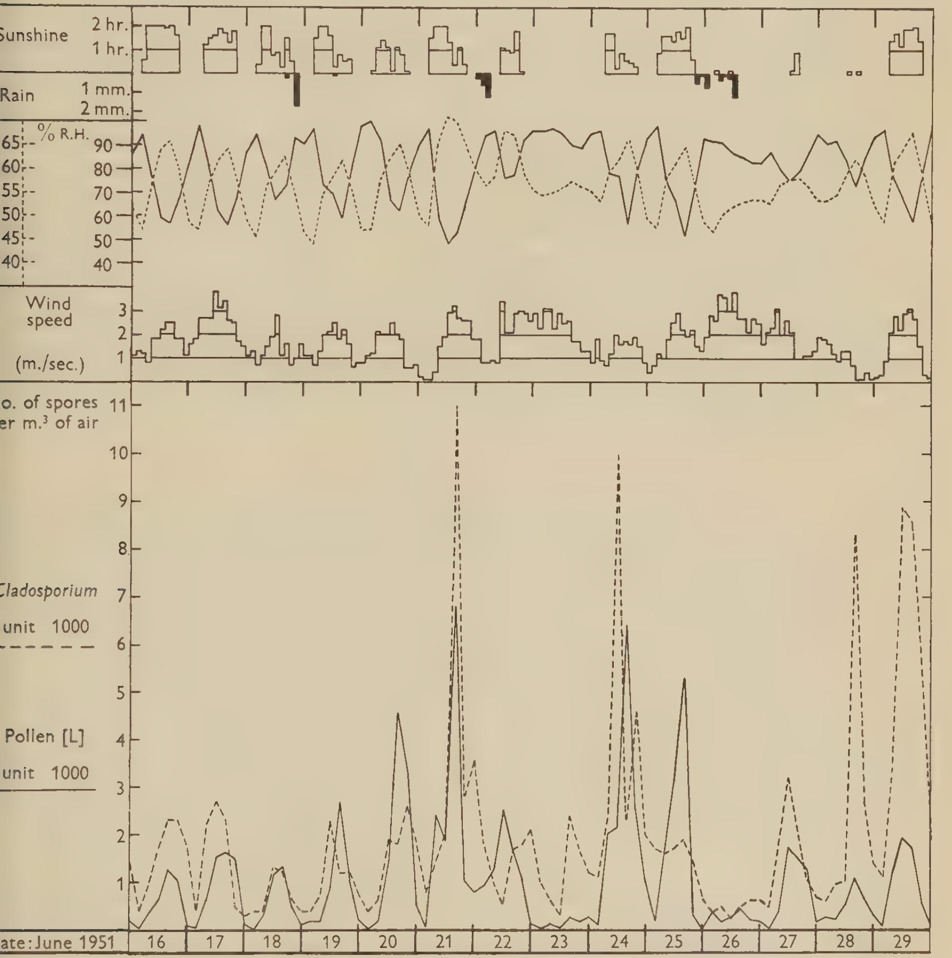
Group counts were made of total pollens, coloured basidiospores, hyaline basidiospores, and uredospores. 'Generic' group counts were made of *Alternaria*, *Cladosporium*, *Erysiphe* and the *Ustilago avenae* group; *Phytophthora infestans* and *Polythrincium trifolii* were identified as species with reasonable certainty. Many hyaline or faintly coloured spores were caught, few of which can be identified at present. From these spores two readily recognizable but artificial composite groups have been separated to indicate changes in the air spora. Group A: composed of filiform spores having a ratio of length to breadth greater than 20 : 1. These are large spores, either septate or aseptate and are probably ascospores (from such genera as *Epichloe* or *Claviceps*) (Pl. 24, fig. 4). Group B: composed of multiseptate fusiform spores characterized by the presence of a markedly swollen cell. These are also probably ascospores (Pl. 24, fig. 4). (Some of this group are slightly coloured and may belong to the genus *Leptosphaeria*.) The remainder of the hyaline spores, not included in any named group, were placed in Group C: 'Hyaline unidentified'. This includes representatives of *Penicillium*, *Botrytis*, many ascospores and splash-dispersed spores. Numerous other coloured spore types including some from Pteridophytes have been recognized, but were usually few and not counted.

RESULTS

The great advantage of the moving slide trap is that it allows changes in the air spora to be closely related to meteorological data. Three periods have been chosen to show how different spore types behave under a variety of weather conditions.

16–29 *June* 1951 (Text-fig. 2)

The first period shows the extent and the constancy of the pattern of diurnal fluctuations in the spore load of total pollen [L] and *Cladosporium*, during a 14-day period in June 1951. *Dactylis glomerata* reached its peak of flowering at the middle of the period and is probably a common constituent of the pollen group at this time. This grass was common in the plot (of about $\frac{1}{8}$ acre) in which the trap was situated, the surrounding crops were mangolds to the east and permanent grass and linseed to the west, from which direction the prevailing wind blows. The upper part of Text-fig. 2 shows sunshine, rainfall and wind velocity (2 m. a.g.l.) plotted at 2-hourly intervals; also temperature and relative humidity (deter-

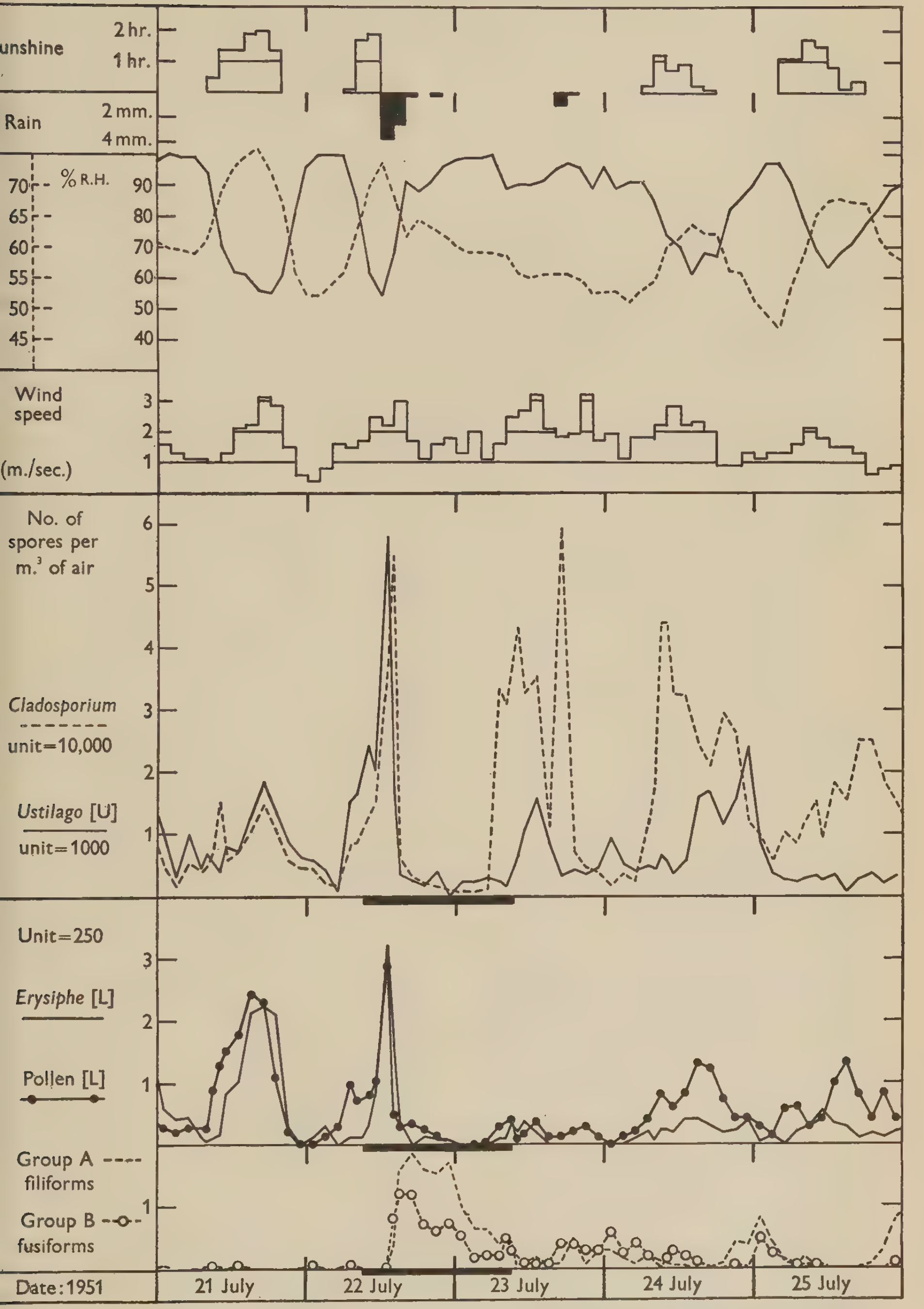


Text-fig. 2. Changes in concentration of air-borne *Cladosporium* spores and pollen in relation to weather. Estimates at 4 hr. intervals between 0001, 16 June 1951 and 2359, 29 June 1951.

mined by mercury-in-steel wet- and dry-bulb thermograph in a Stevenson Screen 4 ft. a.g.l.) plotted at 4-hourly intervals. Pollens and *Cladosporium* are both at their lowest concentration shortly after midnight, when the temperature and wind velocity are low and the relative humidity is high. Maximal concentrations are usually reached in the afternoon when these conditions are reversed. Other spore types, such as *Alternaria*, *Erysiphe* and uredospores, which were less common, behaved similarly. The *Ustilago avenae* group, which occurred in considerable quantity, often had its maximum and minimum several hours later than the types already mentioned, and its periodicity was less regular. The effects of certain combinations of weather are clearly seen, for instance the small amount of pollen on 23, 26 and 28 June was associated with cool dull weather which probably inhibited anthesis. On 23 and 26 June there was also little *Cladosporium*, and its normal periodicity was disturbed. On 28 June *Cladosporium* concentration reached 8300/m.^{3} despite the dullness of the day and small amount of pollen. Rain was never heavy during the period but there is a suggestion that it reduced *Cladosporium* concentrations on 18, 22 and 26 June. On the first two of these days the period of pollen liberation probably missed the rain with the result that spore load of this group was little affected, whereas the more prolonged rain of the morning of 26 June severely limited liberation.

21–25 *July* 1951 (Text-fig. 3)

Alterations in the air spora may sometimes be closely associated with particular weather changes. The second example deals with such a period, when a 7-day spell of warm dry weather ended in a thunderstorm at 1330 on 22 July 1951. During this period the trap was operating in a quarter-acre plot of potatoes, 200 yards north of its previous site. The top part of the figure again shows meteorological data plotted every 2 hr. The lower three panels show the concentrations of six spore types. Up to noon 22 July there was an air spora typical of dry weather and *Cladosporium*, *Ustilago* (U), *Erysiphe* (L) and pollens (L) showed the same type of diurnal periodicity as already described, and there were very few representatives of the hyaline Groups A or B. The thickened part of the time axis shows the period when slide H/86 was exposed (0900 on 22 July to 0900 23 July). The deposit on this slide was strongly zoned because of changes in the weather (Pl. 24, fig. 1). Before the rain the heavy concentration of spores, together with numerous dust and smoke particles, shows on the photograph as a dense deposit on the left-hand side. During the forenoon (8–12 hr.) the load of the dry-air spore types may have increased rapidly because the temperature and wind speed increased and the air became unstable with developing thunder conditions. A photomicrograph of a representative field of the slide at 1300 (Pl. 24, fig. 2), just before the rain, shows conidia of *Erysiphe*, *Alternaria*, *Cladosporium* and spores of a smut, probably *Ustilago perennans*. There are also smoke particles, numerous 'droplets' and a conidiophore. A count at 1400 shows that the concentrations of *Ustilago*, *Erysiphe* and pollens had been reduced to $\frac{1}{3}$, $\frac{1}{2}$ and $\frac{1}{6}$, respectively, of their concentration at the 1300 estimate. By contrast *Cladosporium* had increased from 36,000/m.^{3} at 1300 to 55,000/m.^{3} at 1400.



Text-fig. 3. Changes in concentration of six spore groups in relation to weather, particularly rainfall, between 0001, 21 July 1951 and 2359, 25 July 1951.

This transient increase in *Cladosporium* when rain starts to fall has been noted on other occasions and needs further investigation. On this occasion it may have happened because the thunderstorm air-mass contained a *Cladosporium* cloud, because the wind changed direction, increased in velocity, or possibly because spores were more easily liberated during high humidity conditions. The effect is even more striking at 1700 on 23 July when the rainfall was not accompanied either by thunder, or by a change in wind velocity or direction as shown by a Dines anemobiograph. Ainsworth (1952) has recently found higher numbers of *Cladosporium* colonies on Petri dishes exposed while rain was falling, than on those exposed during dry weather. The changed spora at 1400 cannot be shown effectively by a photograph of a single field, but Pl. 24, fig. 3, does show large numbers of *Cladosporium* spores and a single *Erysiphe* conidium. The reduction in the numbers of *Ustilago* spores is particularly noticeable. The same photomicrograph shows the first appearance of filiform and fusiform spores of Groups A and B, presumably ejected from mature asci after wetting by rain. These changes are reflected in the reduced density of the deposit caught on the slide (Pl. 24, fig. 1), and they become more pronounced with a continuation of the rain. The removal of spores from the air by rain has been confirmed by a different method of trapping (Gregory, Hirst & Last, *unpublished*).

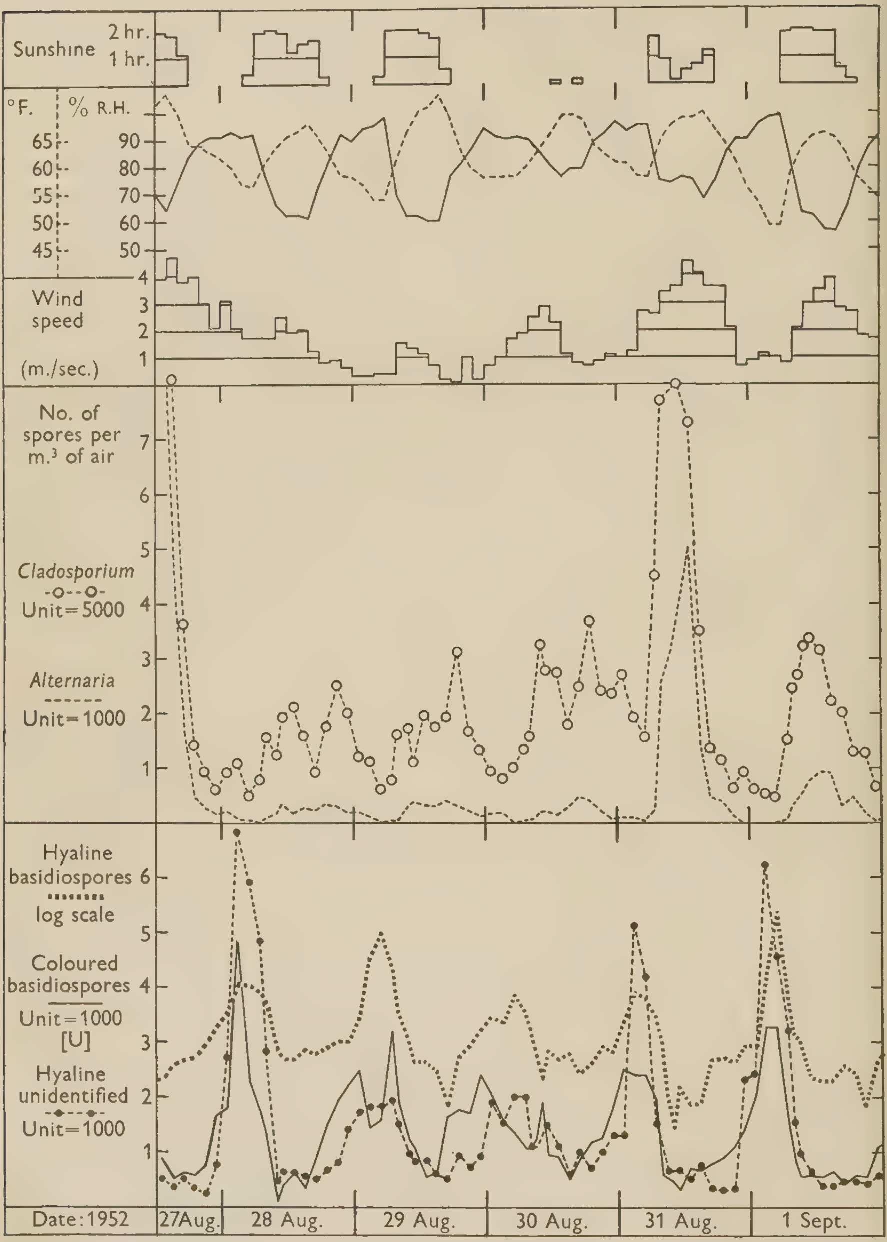
By 1700 on 22 July, a flora characteristic of damp air had replaced that typical of dry air which had existed up to 4 hr. previously (Pl. 24, fig. 4). Variations in the spore load of Groups A and B, both of which reached their peak by 1700, indicate the first stage of this change. Such a rapid liberation may be explained by the fact that many asci had probably matured but had been unable to liberate their spores during the dry weather. Subsequently Groups A and B slowly became less frequent except for brief increases after rain between 1800 and 2000 on 22 July, and between 1600 and 2000 on 23 July, and again with high humidity and possibly dew close to midnight on 24 and 25 July. During late summer and autumn these groups normally occur in greatest numbers at night when the air is usually damp. The effect of rain at approximately 2100 on 22 July was visible on the slide as a temporary decrease in the density of deposit, the larger particles of which at this time were chiefly smoke particles. This second rain shower seemed to have the effect of greatly increasing the numbers of minute unidentified hyaline spores of Group C. Counts for this group are not given, but the photograph (Pl. 24, fig. 5) of a field at 2300 shows very large numbers of spores of this type, and also the first of the hyaline basidiospores which became dominant later. Many of the small hyaline spores of Group C may be splash-dispersed types. The hyaline basidiospores, which are readily identifiable by their apiculus, reached a peak concentration between 0100 and 0300 on 23 July. A single count at about 0200 showed 30,000/m.^{3} and an impression of the number is given in Pl. 24, fig. 6, which shows a field from the slide at 0100 on 23 July. Soon after dawn, there was another rapid change; the hyaline basidiospores disappeared and were replaced by many *Cladosporium* spores (Pl. 24, fig. 7). 23 July was dull and humid, with a slowly declining temperature, and once again pollens were not liberated.

The slowness with which *Erysiphe* recovered may be explained by rain damage to its superficial mycelium (Yarwood, 1936*a*). The last two days shown were dry and had average day temperature. The air spora then returned towards that existing before the thunderstorm, with the addition of a hyaline component probably maintained because the ground was wet.

27 *August*–1 *September* 1952 (Text-fig. 4)

During the third period, in the late summer of 1952, the trap was again situated centrally in a ¼ acre plot of potatoes, 100 yards north of the site first described. This period of 5½ days was of average temperature and included some nights with heavy dew formation. Rainfall is not shown in the text-figure because there was only one light shower of 0·25 mm. between 1000 and 1100 on 30 August. *Cladosporium* and *Alternaria* show the same diurnal periodicity again, although concentrations were low and variable, from 28 to 30 August inclusive. These were days of low wind velocity and during the whole of the period shown there is a suggestion that the concentration of these organisms may have been governed by the occurrence of high wind velocities. However, in the two earlier examples *Cladosporium* does not show this effect consistently. The lowest panel of Text-fig. 4 shows changes of spore load of 'hyaline basidiospores' plotted for convenience on a log scale and also of 'coloured basidiospores' [U], and Group C 'unidentified hyaline spores'. In striking contrast to the diurnal periodicity of *Cladosporium* and *Alternaria* these spores occur in greatest abundance soon after midnight and are least common in the afternoon.

Between 27 August and 1 September records were made of the time of formation and persistence of dew on potato leaves in the crop surrounding the trap. The records may help to explain the violent changes in the occurrence of hyaline basidiospores. Gregory (1952*a*) has suggested that the spores from *Sporobolomyces* spp. (Buller, 1933) growing on leaf surfaces are a large component in the early morning peak load of the group, although 'white'-spored agarics are no doubt also included. There was heavy dew on the night of 27–28 August, despite wind averaging about 2 m./sec., and the peak concentration of hyaline basidiospores was 11,000/m.^{3} at 0300 on 28 August, it is probable that this would have been much greater had the wind velocity been lower. Heavy dew formation in low wind speed conditions occurred on the nights of 28–29 August and 31 August–1 September, when peak concentration of 96,000 and 270,000 hyaline basidiospores per cubic metre occurred at 0500, respectively. Low temperature on the latter night led to mist formation as well as dew. The remaining two nights, 29–30 August and 30–31 August, had only slight and interrupted dew formation and the peak concentrations were 7000 and 8000/m.^{3} at 0500 and 0300, respectively. The maximal concentration of these hyaline basidiospores appears to occur at approximately the time when dew starts to evaporate; the reason for this is not known. As reported by Gregory & Hirst (1952) daily mean concentrations of coloured basidiospores, derived from agarics and polypores, greater than 1000/m.^{3} are common at Rothamsted during the late summer and autumn. This spore group again shows a regular night maximum spore load. Much



Text-fig. 4. Contrasted periodicity of concentration with day- and night-maximum spore groups in relation to weather, in the period 1300, 26 August 1952 to 2359, 1 September 1952.

published work (Buller, 1909; Hilborn, 1942) suggests that the rate of spore liberation is little affected by diurnal changes in temperature, humidity or light intensity. But many of the methods used are unsuitable for accurate quantitative work for various reasons, notably imperfect control of slow air movements, and the question requires re-investigation in controlled conditions in a wind tunnel. If the rate of liberation is approximately constant, changes in the spore load may depend on the extent of dilution of the spore cloud by turbulence. At night thermal turbulence is low and the turbulence generated by obstacles is frequently reduced owing to lower wind speed; thus higher concentrations would be expected at night. The spores included in Group C, unidentified hyaline, are probably derived from a variety of substrates and fructifications and become air-borne by various processes. In addition different types may be dominant on different nights or at different times on the same night. It is therefore not surprising that it is more difficult to correlate their behaviour with any one weather factor such as the existence of dew. The difficulty of interpreting these results is emphasized by the similarity of the diurnal periodicity in these three night-maximum spore groups, despite the probability that with each group the causes of the variations are quite different.

Diurnal periodicity

These three examples show how rapidly and extensively the air spora can vary; also that a diurnal periodicity is usual with some spores, but can be modified. Changes in wind velocity, temperature, humidity, sunshine, rainfall or dew are clearly responsible for some modifications of diurnal periodicity. Another frequent cause, so far mentioned only briefly, is wind direction. Some of the spores trapped may have travelled long distances, and it is usually impossible to relate the number of spores caught to known sources in the vicinity of the trap. For an organism of limited distribution this difficulty may be overcome by siting the trap at the centre of a source sufficiently powerful to swamp the small numbers arriving from distant sources. It was for the study of the spores of one such pathogen, *Phytophthora infestans*, that the trap was placed centrally in potato plots during the period of the potato blight epidemic. Another method of reducing the effect of wind direction on diurnal periodicity, is to consider the mean periodicity over as long a period as possible. Assuming that, as at Rothamsted, wind direction and time of day are not correlated this method should give reliable averages. Such averages may be made either for all days, or only for dew, rain or dry days.

In Text-fig. 5 such mean periodicity curves are shown for thirteen spore groups. The curves were derived from the geometric means of the estimated number of spores per cubic metre of air at each of the times of estimation. The values were then converted into a percentage of the highest of these geometric means. This method of plotting allows easy comparison of the periodicity of various spore types, the actual values of the geometric means have little interest unless considered in relation to comprehensive data on seasonal fluctuations of spore load. The number of days on which the mean is based is smaller than desirable for some types, and is stated in the legend to the figure. The days included have not been selected for any particular type of weather; they include days with high and low concentrations of spores and days at different times in the season of liberation.

Pollens, Cladosporium and Alternaria. These three groups are all important allergens and behave similarly, with well-defined maximum concentrations recurring in the afternoon. The periodicity of particular pollen types derived from such plants as grasses and plantains (Hyde & Williams, 1945, 1946) will be masked by the heterogeneity of the total pollen group. Pollen grains with diameter greater than 20μ show a subsidiary peak at 0400 during the period shown in Text-fig. 2, and this may come from *Dactylis glomerata* and other grasses, which usually flower early in the day. The results agree with those of Hyde & Williams concerning the effects of weather. Low concentrations occur regularly at night and in cool, damp weather, rain rapidly removes air-borne pollens but they are quickly replaced if conditions become suitable for the rupture of anthers.

Few *Alternaria* spores were caught throughout 1951; many more were caught in 1952, but they were at all times much scarcer than *Cladosporium* spores. This is at variance with surveys in America, where *Alternaria* has often been found to be more common than *Cladosporium*. The better sampling obtained with the suction traps may partially account for this, as a large proportion of the *Cladosporium* spores are less than 10μ in diameter and would not be efficiently caught by the older types of spore trap. However, this cannot be the whole reason because 'gravity slides', vertical cylinders or Petri dishes exposed at Rothamsted over a number of years also catch more *Cladosporium* than *Alternaria*. Clumps of *Cladosporium* spores appear to be most common before the daily peak concentration is reached and when rain starts; clumping is least common after prolonged rain and at night.

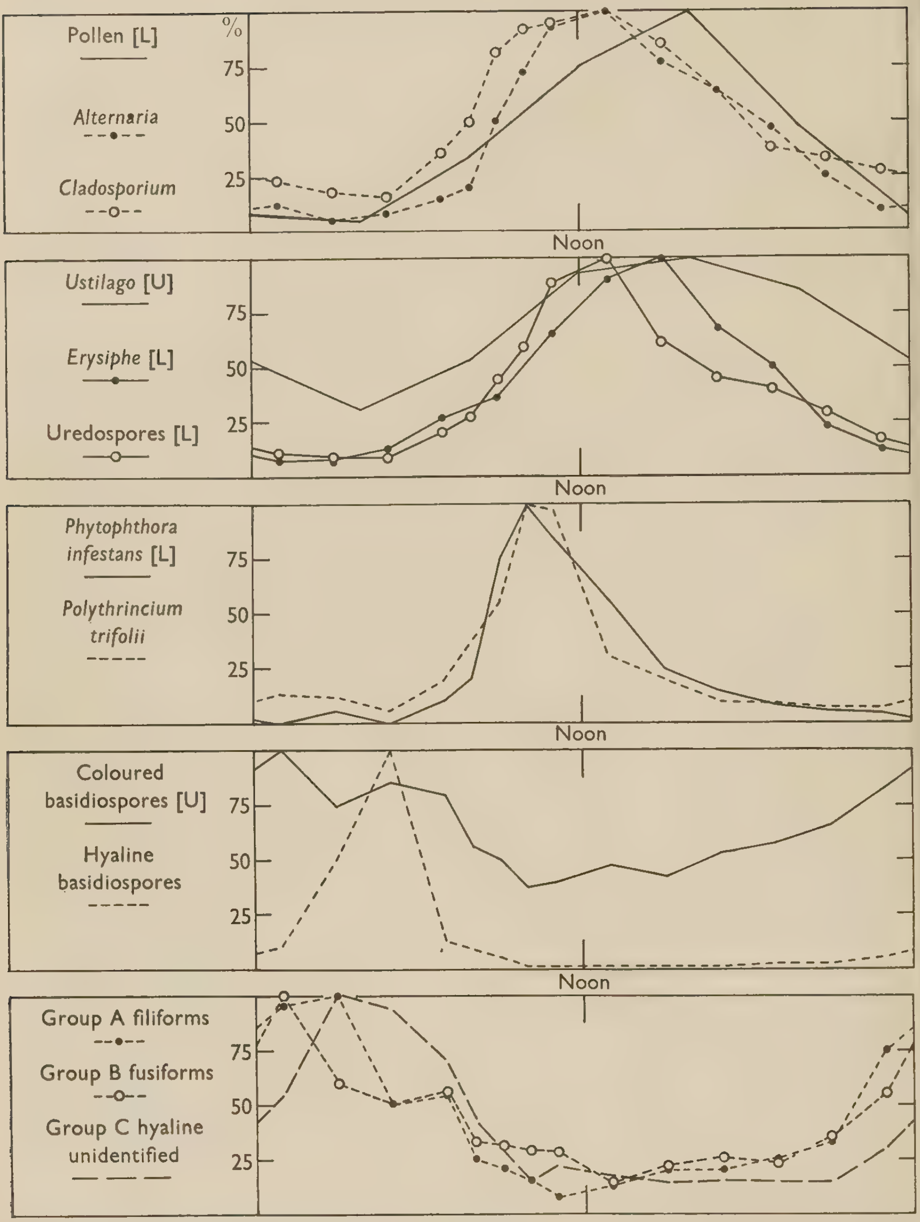
Ustilago, Erysiphe and uredospores. In this group of plant pathogens two of the spore types have also been claimed as allergens (Cadman, 1924; Wittich, 1939). *Erysiphe* is a common and important genus of obligate plant parasites without any known importance as allergens. Yarwood (1936*b*) has shown that light controls the periodicity of spore formation in *E. polygoni*, a minor component of our *Erysiphe* group. In natural conditions he found that conidia were abstricted only between 0800 to 1200, and he thought liberation then depended on external forces. With slides exposed for 4 hr. periods, Yarwood found that spore concentrations were greatest about noon, soon after the time of maximal maturation. This agrees with catches in the suction trap. It is unfortunate that morphological differences prevented the extension of Yarwood's work to *E. graminis*, the major component of the *Erysiphe* group, but the time of the maximum catches suggests that it behaves similarly to *E. polygoni*.

The uredospore group was dominated by the rusts of wheat and barley, although a considerable variety of species was present in small numbers. Teleutospores were caught occasionally but no aecidiospores were recognized. No active discharge mechanism is known for uredospores and it is interesting to see how closely their periodicity curve resembles those of *Alternaria* and *Cladosporium*.

Once they become exposed on the diseased inflorescence, the number of *Ustilago* spores capable of dispersal on any particular day depends very little on the activities of the fungus or its host plant. The periodicity observed must be determined chiefly by the varying ease with which spores are detached from the spore mass, and by atmospheric conditions such as wind velocity and turbulence. Spore clumps are common and their numbers vary in a similar way to that described for *Cladosporium*. Clumping may partially explain the ease with which *Ustilago* spores are removed from the air by rain in comparison with coloured basidiospores of similar size.

Phytophthora infestans and Polythrincium trifolii. These species exhibit a periodicity different from any other of the groups studied. Its regularity suggests either a definite discharge mechanism or the daily recurrence of atmospheric conditions favouring the liberation of spores. In *Phytophthora* the same rhythm is followed at a height of 2 and 0·5 m. The rapid liberation of sporangia starts at about 0800 and the peak is reached at 1000. There were a few days when a few spores were found later than 1900 or earlier than 0700 but these were exceptional. Rain and strong winds apparently modify this periodicity slightly but seem not to be the major controlling factors. The start of the liberation and the time of the peak concentration would both be consistent with de Bary's (1887) suggestion that spore discharge occurs with the hygroscopic twisting movements of sporangiophores exposed to rapidly changing humidity. This method, and forcible ejection from the sterigmata of isolated sporangiophores under conditions of decreasing vapour pressure, have both been claimed as discharge mechanisms in *Peronospora tabacina* (Pinckard, 1942). There is evidence that the sporulation processes of *Phytophthora infestans* are not affected by light (Crosier, 1934) as they are said to be in other downy mildews (Yarwood, 1937). Nevertheless the peak concentration seems to occur at the same time as that quoted by Yarwood for *Pseudoperonospora humuli*. The spores of *Phytophthora infestans* are easily killed by drying and germinate most rapidly in water droplets (Crosier, 1934). Liberation coincides with rising temperature and the disappearance of dew and rain-drops from the leaves of potential hosts. The very close similarity of the periodicity curve for *Polythrincium trifolii* suggests that this species may also depend on similar diurnal weather changes for liberation.

Coloured and hyaline basidiospores. In contrast to all the groups already described, both coloured and hyaline basidiospores reach their maximum concentration during the early hours of the morning, and are least common during daylight. The two basidiospore groups differ greatly from each other in the amplitude of their diurnal periodicity; the factors that may control this have already been discussed. Spore clumps are rare, because each spore is liberated individually from its sterigma. The rapidity with which hyaline basidiospores disappear is one of the most dramatic of the short-period changes that occur in the air spora. On 1 September 1952, for instance, within 4 hr. the concentration dropped from over a quarter of a million to about a thousand per cubic metre. At this time of day their disappearance is unlikely to be due to deposition and it is interesting to speculate on their fate. The rapid decrease in hyaline basidiospore concentration after dawn may be the result of increased turbulence dispersing throughout the air to considerable altitudes, a dense cloud formed close to the ground.



Text-fig. 5. Mean diurnal periodicity curves of thirteen spore groups expressed as percentage of the peak geometric mean concentration. Number of days included in the mean for each group: pollen (36), *Alternaria* (24), *Cladosporium* (25), *Ustilago* (26), *Erysiphe* (25), uredospores (28), *Phytophthora infestans* (31), *Polythrincium trifolii* (12), coloured basidiospores (21), hyaline basidiospores (7), Group A (24), Group B (25) and Group C (6).

Groups A, B and C. These groups are at present very imperfectly understood, but contain important plant pathogens. The mean periodicity curves all show maxima at night, although this pattern can be greatly altered on wet days. The peaks of the curves also suggest a succession of dominant hyaline spore types. The heterogeneous Group C contains a further pronounced succession, starting before midnight with septate ascospores, followed by minute rod-shaped or jointed spores in the early morning, while just before dawn there appear a few spores which may belong to Entomophthoraceae. During daylight, unidentified downy mildews, *Botrytis cinerea*, *Penicillium* spp. and other types, occur and have been included in Group C.

Discussion

The use of an apparatus which traps, with nearly uniform efficiency, spores of a wide range of size allows a comprehensive study of the complete air spora, and shows the deficiencies of estimates made by earlier methods. Thus 'hyaline basidiospores', which were very rarely seen during several years' work with freely exposed surface traps, were the group which reached the highest concentration during the sample periods quoted. Although this is admittedly a composite group, its maximum concentration was more than three times as great as that reached in the same period by *Cladosporium*, its nearest rival. The separation, on a moving slide, of spores trapped at different times allows the atmospheric spore load to be estimated at frequent intervals throughout the growing season. This shows that the air spora is continuously changing, both qualitatively and quantitatively, a constant spore load for more than a few hours is exceptional.

Routine suction spore trapping should aid the recognition of additional air-borne allergens. It can improve the methods of assessing daily mean spore concentration and provide, for the first time, information on the brief maximum concentrations to which patients are exposed. The difficulty of interpreting results and the magnitude of the changes in the numbers of air-borne spores make it clear that neither daily exposure of horizontal slides nor the exposure of single Petri dishes, for a few minutes each day, can be expected to provide this information.

The new methods offer mycologists and plant pathologists a means of extending spore trapping studies to groups of organisms with which previous methods have been of little value. Correlating changes of spore load with frequent and detailed weather records, both in and above crops, can provide much information about the biology of the fungi and epidemiology. No doubt much modification of the present methods will be required when they are applied to new problems. Trapping in the open air will have to be adjusted for the study of particular organisms, and be supported by laboratory experiments using partially controlled environments, before the effects of different conditions can be fully assessed. Only exceptionally do these preliminary studies indicate the factors or interactions of factors controlling the observed fluctuations in spore load. As Gregory (1952*b*) has pointed out, these controls are both biological and physical and may operate at many points in the dissemination cycle. The formation of spores has long been studied, but many more quantitative data are desirable. Similarly, although much attention has been paid to the more dramatic mechanisms whereby spores are actively discharged, it remains unknown how and in what conditions the spores of many common and important allergens and pathogens are liberated. Spores, when formed and past the obstacle of 'take off', suffer the dangers of involuntary transport through the air, including the possible loss of viability, and then the difficulty of once again penetrating the atmospheric boundary layer surrounding a suitable substrate or host. Only through knowledge of all these processes can we hope to understand the dynamic quality of the air spora.

REFERENCES

Ainsworth, G. C. (1952). The incidence of air-borne *Cladosporium* spores in the London region. *J. gen. Microbiol.* **7**, 358–361.

Alvarez, J. C. & Castro, J. F. (1952). Quantitative studies of air-borne fungi of Havana in each of the 24 hours of the day. *J. Allergy*, **23**, 259–264.

Buller, A. H. R. (1909). *Researches on the fungi*, vol. 1. London: Longmans, Green and Co.

Buller, A. H. R. (1933). *Researches on the fungi*, vol. 5. London: Longmans, Green and Co.

Cadman, F. T. (1924). Asthma due to grain rusts. *J. Amer. med. Ass.* **83**, 27.

Crosier, W. (1934). Studies in the biology of *Phytophthora infestans* (Mont.) de Bary. *Mem. Cornell agric. Exp. Sta.* **155**, 40 pp.

De Bary, A. (1887). *Comparative morphology and biology of the fungi, mycetozoa and bacteria.* Clarendon Press, Oxford.

Downs, W. G. (1943). Polyvinyl alcohol: a medium for mounting and clearing biological specimens. *Science*, **97**, 539.

Durham, O. C. (1944). The volumetric incidence of atmospheric allergens: II. Simultaneous measurements by volumetric and gravity slide methods. Results with Ragweed pollen and *Alternaria* spores. *J. Allergy*, **15**, 226–235.

Gregory, P. H. (1951). Deposition of air-borne *Lycopodium* spores on cylinders. *Ann. appl. Biol.* **38**, 357–376.

Gregory, P. H. (1952*a*). Spore content of the atmosphere near the ground. *Nature, Lond.*, **170**, 475.

Gregory, P. H. (1952*b*). Presidential Address: Fungus spores. *Trans. Brit. mycol. Soc.* **35**, 1–18.

Gregory, P. H. & Hirst, J. M. (1952). Possible role of basidiospores as air-borne allergens. *Nature, Lond.*, **170**, 414.

Gregory, P. H., Hirst, J. M. & Last, F. T. (Unpublished.)

Gregory, P. H. & Stedman, O. J. (1953). Deposition of air-borne *Lycopodium* spores on plane surfaces. *Ann. appl. Biol.* (In the press.)

Hawes, R. C., Small, W. S. & Miller, H. (1942). An apparatus for determining the pollen content of air and notes on pollen survey method. *J. Allergy*, **13**, 474–487.

Heise, H. A. & Heise, E. R. (1950). Meteorologic factors in the distribution of pollens and molds. *Ann. Allergy*, **8**, 641–644 and 681.

Hilborn, M. T. (1942). The biology of *Fomes fomentarius*. *Bull. Maine agric. Exp. Sta.* **409**, 161–214.

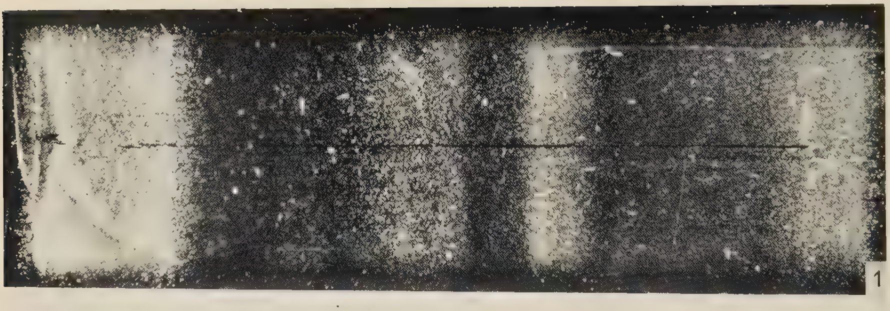
Hirst, J. M. (1952). An automatic volumetric spore trap. *Ann. appl. Biol.* **39**, 257–265.

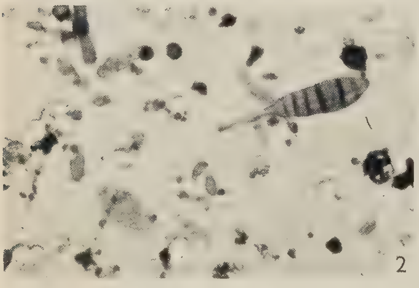
Hyde, H. A. & Williams, D. A. (1945). Studies in atmospheric pollen: II. Diurnal variation in the incidence of grass pollen. *New Phytol.* **44**, 83–94.

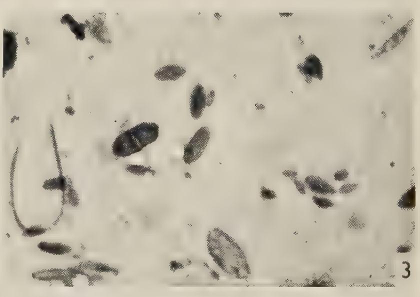
Hyde, H. A. & Williams, D. A. (1946). Studies in atmospheric pollen: III. Pollen production and pollen incidence in Ribwort Plantain (*Plantago lanceolata* L.). *New Phytol.* **45**, 271–277.

Jones, M. T. (1952). Time of day of pollen shedding of some hay-fever plants. *J. Allergy*, **23**, 247–258.

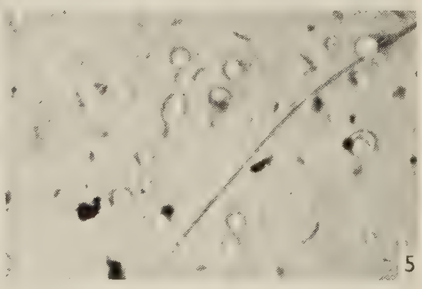
Keitt, G. W. & Jones, L. K. (1926). Studies of the epidemiology and control of apple scab. *Res. Bull. Wis. agric. Exp. Sta.* **73**, 104 pp.

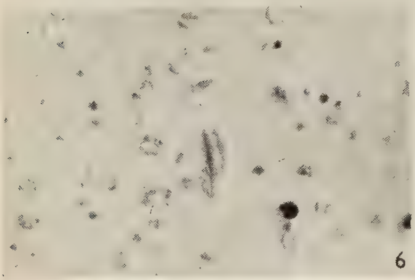

1

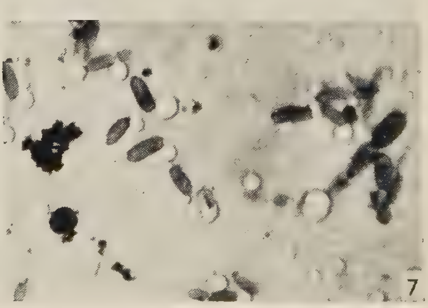

2


3

4


5


6


7

Landahl, H. D. & Herrmann, R. G. (1949). Sampling of liquid aerosols by wires, cylinders and slides, and the efficiency of impaction of the droplets. *J. Colloid Sci.* **4**, 103–136.

May, K. R. (1945). The Cascade Impactor: An instrument for sampling coarse aerosols. *J. sci. Instrum.* **22**, 187–195.

Pinckard, J. A. (1942). The mechanism of spore dispersal in *Peronospora tabacina* and certain other Downy Mildew fungi. *Phytopathology*, **32**, 505–511.

Wittich, F. W. (1939). Further observations on allergy to Smuts. *Lancet*, **49**, 382.

Yarwood, C. E. (1936*a*). The tolerance of *Erysiphe polygoni* and certain other Powdery Mildews to low humidity. *Phytopathology*, **26**, 845–859.

Yarwood, C. E. (1936*b*). The diurnal cycle of the powdery mildew *Erysiphe polygoni*. *J. agric. Res.* **52**, 645–657.

Yarwood, C. E. (1937). The relation of light to the diurnal cycle of sporulation of certain Downy Mildews. *J. agric. Res.* **54**, 365–373.

EXPLANATION OF PLATE 24

Fig. 1. Photograph by reflected light of deposit on Slide H/86 (exposed 0900 22 July 1951, left-hand side, to 0900 23 July 1951).

Figs. 2–7. Photomicrographs of representative fields from Slide H/86 at approximately 1300, 1400, 1700, 2300, 0100, 0700 respectively. (Mag. × 400.)

(*Accepted for publication* 15 *December* 1952)

LIST OF MEMBERS

Corrected to 13 July 1953

Honorary Members

Barnes, B., D.Sc., Ph.D., F.L.S., 19 High Park Road, Kew Gardens, Richmond, Surrey. (1922.) (1950.)

Dodge, Dr B. O., New York Botanical Garden, Bronx Park, New York 58, N.Y., U.S.A. (1946.)

Falck, Professor R. (1946.)

Gäumann, Professor E., Eidgenössischen Technischen Hochschule, Universitätstrasse, Zürich, Switzerland. (1946.)

Heim, R., Sous-Directeur au Muséum d'Histoire Naturelle, 11 Rue de Médicis, Paris (6^e), France. (1930.) (1946.)

Moore, W. C., M.A., Ministry of Agriculture, Plant Pathology Laboratory, Milton Road, Harpenden, Herts. (1922.) (1951.)

Pearson, A. A., F.L.S., Nutcombe House, Hindhead Road, Hindhead, Surrey. (1911.) (1946.)

Ramsbottom, J., O.B.E., D.Sc., M.A., F.L.S., British Museum (Natural History), Cromwell Road, South Kensington, London, S.W. 7. (1910.) (1945.)

Swanton, E. W., O.B.E., A.L.S., Museum House, High Street, Haslemere, Surrey. (1898.) (1947.)

Wakefield, Miss E. M., O.B.E., M.A., F.L.S., 20 Castlegate, Richmond, Surrey. (1911.) (1941.)

Watson, W., D.Sc., A.L.S., Cedene, 274 Cheddon Road, Taunton, Somerset. (1923.) (1951.)

Members

Aaron, J. R., c/o The Forestry Commission, 25 Savile Row, London, W. 1. (1952.)

Acheson, Miss M. E., Ph.D., Department of Botany, University College, Dundee. (1949.)

Adams, Rev. J. H., Landulph Rectory, Hatt, Saltash, Cornwall. (1919.)

Ainsworth, G. C., B.Sc., Ph.D., F.L.S., Department of Botany, Hatherly Laboratories, University College of the South West, Prince of Wales Road, Exeter. (1931.)

Alaily, Dr Y. A. S. El, Société Anonyme de Wadi Kom-Ombo, P.O. Box 738, Cairo. (1935.)

Alcock, Mrs N. L., M.B.E., F.L.S., 108 Colebrook Street, Winchester, Hants. (1919.)

Allchin, J. P., A.I.C. (1942.)

Allen, C. Winckworth, 4 Leinster Square, Rathmines, Dublin, Eire. (1946.)

Alsford, Miss J., 38 Cotesbach Road, London, E. 5. (1950.)

Andersson, O., Botaniska Museet, Box 5, Lund, Sweden. (1947.)

Angus, A., Forest Herbarium, Department of Forestry, Imperial Forestry Institute, The University, Oxford. (1949.)

Apinis, A., Department of Botany, The University, Nottingham. (1949.)

de Arêa Leão, A. E., Mycology Laboratory, Instituto Oswaldo Cruz, Caixa Postal 926, Rio de Janeiro, Brazil. (1950.)

Arif, A. G., M.A., M.Sc., Ph.D., Assistant Professor of Botany, Agricultural College, Lyallpur, West Punjab, Pakistan. (1948.)

Ashour, W. El., Ph.D., Lecturer, Faculty of Agriculture, Shebein El Kom, Egypt. (1946.)

Atkins, F. C., Yaxley, Peterborough, Northants. (1945.)

Austwick, P. K. C., Department of Botany, Hatherly Laboratories, University College of the South West, Prince of Wales Road, Exeter. (1946.)

Aytoun, R. S. C., 18 Leyton Paddock, Stoke Poges, Bucks. (1950.)

Badcock, E. C., Forest Products Research Laboratory, Princes Risborough, Bucks. (1944.)

Baker, Professor K. F., University of California, 405 Hilgard Avenue, Los Angeles 24, California, U.S.A. (1946.)

Baker, R. E. D., B.A., Imperial College of Tropical Agriculture, Trinidad. (1944.)

Balfour-Browne, Mrs F. L., M.Sc., Department of Botany, British Museum (Natural History), Cromwell Road, South Kensington, London, S.W. 7. (1930.)

Banbury, G. H., B.Sc., Department of Botany, University College, Ibadan, Nigeria, British West Africa. (1949.)

Bannan Miss L., B.Sc., A.R.C.S., 8 Belgrave Road, Chorlton cum Hardy, Manchester. (1945.)

Barnes, Miss E. M., B.Sc., Sir W. Dunn School of Pathology, University of Oxford. (1943.)

Barrington, Dr F. J. F., 14A Upper Wimpole Street, London, W. 1. (1901.)

Bates, G. R., Ph.D., c/o Division of Agriculture and Lands, Branch of Botany and Plant Pathology, P.O. Box 100, Causeway, Salisbury, Southern Rhodesia. (1930.)

Batista, Professor A. Chaves, Instituto de Pesquisas Agronomicas, Dois Irmãos, Recife, Pernambuco, Brazil. (1950.)

Batko, Dr S., Alice Holt Lodge, Forestry Commission, Wrecclesham, nr. Farnham, Surrey. (1944.)

Batts, C. C. V., B.Sc., Ph.D., National Institute of Agricultural Botany, Huntingdon Road, Cambridge. (1946.)

Bawden, F. C., M.A., F.R.S., Rothamsted Experimental Station, Harpenden, Herts. (1941.)

Beaumont, A., M.A., Block 2, Government Buildings, Lawnswood, Leeds 6. (1924.)

Beedham, Mrs M., B.Sc., Ystwyth, Godstone Road, Bletchingly, Surrey. (1948.)

Beevor, M. B., Hazel Croft, Cheveley, Newmarket, Suffolk. (1952.)

Bels, Dr P., P.O. Box 558, Waterloo, Quebec, Canada. (1949.)

Bennett, A. J., 84 Henhurst Hill, Burton-on-Trent, Staffs. (1948.)

Bennett, Mrs H. Sterndale, Oaken Wood, Wateringbury, Kent. (1950.)

Bennett, Miss M., B.Sc.(Hort.), East Malling Research Station, nr. Maidstone, Kent. (1945.)

Bewley, W. F., C.B.E., D.Sc., Experimental and Research Station, Cheshunt, Herts. (1922.)

Biggs, Miss R., B.Sc., Ph.D., M.B., B.S., The Radcliffe Infirmary, Oxford. (1945.)

Bilton, P. A., B.Sc., M.Sc., Cambridgeshire High School for Boys, Hills Road, Cambridge. (1950.)

Biraghi, Dr Antonio, Professor of Forest Pathology, Istituto di Patologia Forestale e Agraria, Facoltà Agraria e Forestale, Università degli Studi, Piazzale delle Cascine, Firenze, Italia. (1953.)

Birkinshaw, J. H., D.Sc., Firgrove, 4 Nower Hill, Pinner, Middlesex. (1946.)

Bisby, G. R., Ph.D., Commonwealth Mycological Institute, Ferry Lane, Kew, Surrey. (1921.)

Blackman, Professor V. H., M.A., Sc.D., F.R.S., F.L.S., 17 Berkeley Place, Wimbledon, London, S.W. 19. (1900.)

Blackwell, Miss E. M., M.Sc., F.L.S., Woodsome Lees, Kirkburton, nr. Huddersfield, Yorks. (1917.)

Blumer, Dr S., Schoenenbergstrasse 85, Wädenswil, Switzerland. (1936.)

Boardman, Mrs S., 109 Monkhams Avenue, Woodford Green, Essex. (1950.)

Bond, T. E. T., Ph.D., F.L.S., Horticultural Science Laboratories (University of Bristol), Bracken Hill, Leigh Woods, Bristol, 8. (1945.)

Bottomley, T. L. C., 23 Rygate Crescent, Sheffield, 10. (1950.)

Boughey, Professor A. S., B.Sc., Ph.D., University College of the Gold Coast, Achimota, Gold Coast, West Africa. (1945.)

Boyd, A. E. W., B.Sc., Ph.D., East of Scotland College of Agriculture, 13 George Square, Edinburgh. (1947.)

Bradley Jones, J., N.A.A.S., Plant Pathology Department, University College, Cathays Park, Cardiff. (1948.)

Brady, Miss B. L., B.Sc., Swanmead, Datchet, Bucks. (1948.)

Brenchley, G. H., M.A., N.A.A.S., Staplake Mount, Star Cross, Devon. (1925.)

Brett, Miss M., M.Sc., Ph.D., F.L.S., Greentiles, St Andrews Road, Bridport, Dorset. (1921.)

Brian, P. W., M.A., Ph.D., Sc.D., Imperial Chemical Industries Ltd., Butterwick Laboratories, The Frythe, Welwyn, Herts. (1944.)

Brierley, J. K., 25 Lilac Avenue, Kingsway, Derby. (1951.)

Brind, Mrs J. E., Rothamsted Experimental Station, Harpenden, Herts. (1948.)

Broadfoot, Miss J., B.Sc. c/o Lloyd's Bank, Staines, Middlesex. (1945.)

Brodie, H. J., Ph.D., Department of Botany, Indiana University, Bloomington, Indiana, U.S.A. (1952.)

Brook, M., B.Sc., M.I.Biol., 16 Mottram Road, Chilwell, Notts. (1952.)

Brooke, John, 'Rowmore', Leigh Hill Road, Cobham, Surrey. (1947.)

Brookfield, Miss E., 28 Fourth Avenue, Linden, Johannesburg, South Africa. (1946.)

Brooks, J. S., The Research Department, H. W. Carter & Co., Ltd., The Royal Forest Factory, Coleford, Glos. (1951.)

Brown, Miss A. H. S. 28 Abinger Road, Bedford Park, London, W. 4. (1950.)

Brown, Miss J. C., B.Sc., Highborn Eaves, Effingham, nr. Leatherhead, Surrey. (1952.)

Brown, Professor W., M.A., D.Sc., F.R.S., Imperial College of Science and Technology, South Kensington, London, S.W. 7. (1922.)

Buchwald, Professor N. F., Royal Agricultural and Veterinary College, Rolighedsvej 23, Copenhagen V, Denmark. (1946.)

Buckley, W. D., 'St Anthony', Leigh Park, Datchet, Bucks. (1916.)

Buddin, W., M.A., N.A.A.S., 'Chiltern Court', St Peter's Avenue, Caversham, Reading, Berks. (1921.)

Bull, Miss P., 14 Gresham Road, Hounslow, Middlesex. (1952.)

Bulloch, M. G., Blandford Farm Products, Upper Shaftesbury Road, Blandford, Dorset. (1949.)

Bundy, J. W., B.Sc., Food Research Department, Colworth House, Sharnbrook, Bedford. (1951.)

Bunting, R. H., F.L.S., 3 Stanton Court, Weymouth. (1921.)

Burges, Professor N. A., Department of Botany, The University, Liverpool. (1935.)

Burnett, J. H., M.A., Department of Botany, The University, Oxford. (1948.)

Butler, F. C., B.Sc.Agr., Ph.D., Biological Branch, Department of Agriculture, Box 36, G.P.O., Sydney, N.S.W. (1951.)

Butterworth, D., Ministry of Supply, Tropical Testing Establishment, Port Harcourt, Nigeria. (1950.)

Buxton, E. W., Botany School, Downing Street, Cambridge. (1952.)

Byford, W. J., 72 Oakdale Road, Leystonstone, London, E. 11. (1951.)

Byrde, R. J. W., B.Sc., Ph.D., Homeleigh, Downside, West Town, nr. Bristol. (1952.)

Caldwell, Mrs B. J., B.Sc., Department of Cryptogamic Botany, The University, Manchester, 13. (1943.)

Caldwell, Professor J., D.Sc., Ph.D., Department of Botany, Hatherly Laboratories, University College of the South West, Prince of Wales Rd., Exeter. (1932.)

Caldwell, R., 31 Orient Road, Salford 6, Lancs. (1950.)

Callen, E. O., B.Sc., Ph.D., F.L.S., Department of Plant Pathology, Macdonald College, McGill University, Quebec, Canada. (1941.)

Campbell, A. H., B.Sc., Ph.D., Glaxo Laboratories Ltd., Fermentation Research Division, Sefton Park, Stoke Poges, Bucks. (1934.)

Campbell, W., B.Sc. (Agr.)., N.A.A.S., Staplake Mount, Starcross, Exeter, Devon. (1951.)

Cannons, C. E., 33 Langley Oaks Avenue, Sanderstead, Surrey. (1951.)
Canter-Lund, Dr Hilda, Freshwater Biological Association, The Ferry House, Far Sawrey, Ambleside, Westmorland. (1944.)
Carr, A. J. H., B.Sc., Welsh Plant Breeding Station, Aberystwyth. (1949.)
Carrothers, E. N., Ulster Transport Authority, 21 Linenhall Street, Belfast, Northern Ireland. (1925.)
Cartwright, K. St G., M.A., F.L.S., The Old Vicarage, Towersey, Thame, Oxon. (1913.)
Catsimbas, C., B.Sc., 4 Castellorizon Street, Athens, Greece. (1947.)
Caulton, D. E., 45 Ash Grove, Headingley, Leeds 6. (1949.)
Cavett, J. J., B.Sc., Department of Agriculture, The University, Leeds 2. (1951.)
Cayley, Miss D. M., Foxhall Cottage, Kelshall, nr. Royston, Herts. (1913.)
Chapman, F. P., P.O. Box 1308, Cape Town, South Africa. (1952.)
Charlton, Mrs K. M., Four Oaks, Billingbear Lane, Binfield, Berks. (1945.)
Cheal, W. F., Quarry Bank, London Road, West Malling, Kent. (1927.)
Chesters, Professor C. G. C., B.Sc., M.Sc., Ph.D., Department of Botany, The University, Nottingham. (1930.)
Chowdhury, S., Government Mycologist, Jorhat, Assam, India. (1949.)
Christensen, Dr C. M., University Farm, St Paul 8, Minnesota, U.S.A. (1945.)
Ciferri, Professor Dr R., Laboratorio Crittogamico, Casella Postale 165, Pavia, Italy. (1926.)
Cleary, J. P., N.A.A.S., Block 2, Government Buildings, Lawnswood, Leeds 6. (1948.)
Cleasby, W. B., Lower Honeydon Farm, nr. Bedford. (1949.)
Cleland, J. B., M.D., Professor of Pathology, University of Adelaide, South Australia. (1918.)
Colhoun, J., B.Sc., M.Agr., Ph.D., D.I.C., Plant Disease Division, Ministry of Agriculture, Queen's University, Belfast, Northern Ireland. (1943.)
Collett, Mrs F. W. M., Townsend Farm, Littleton Drew, Chippenham, Wilts. (1948.)
Collinge, A. C., 2 Wood View, Savile Park, Halifax, Yorks. (1951.)
Colombo, Mrs G. M., 32 Carlyle Road, Staines, Middx. (1950.)
Cone, Mrs G. B., Ph.D., 3 Steep Street, Dunedin, C 2, New Zealand. (1951.)
Cooke, Mrs A. S., B.Sc., F.L.S., 20 Grand Avenue, Hassocks, Sussex. (1938.)
Cooper, Mrs V. A. (1921.)
Corner, E. J. H., M.A., F.L.S., Botany School, Downing Street, Cambridge. (1924.)
Cornford, C. E., B.Sc., A.R.C.S., Ph.D., 149 Monks Road, Lincoln. (1953.)
Cotton, A. D., O.B.E., V.M.H., F.L.S., Old Weavers, Pitchcoombe, nr. Stroud, Glos. (1902.)
Cottrell, A. R., B.Sc.(Agric.), 245 Burton Road, West Didsbury, Manchester, 20. (1948.)
Cousins, P., 'Westward', Arthur Road, Wokingham, Berks. (1951.)
Cox, Miss B. J., B.Sc., Elm Lodge, Englefield Green, Surrey. (1953.)
Craig, J. K., Craigs Ltd., The Marsh, Widnes. (1952.)
Critopoulos, Dr P. D. University of Athens, 104 Solonos Street, Athens, Greece. (1949.)
Crosse, J. E., B.Sc., Research Station, East Malling, nr. Maidstone, Kent. (1948.)
Crosse, R., 'The Brambles', 12 Sefton Paddock, Stoke Poges, Bucks. (1951.)
Crowdy, S. H., Imperial Chemical Industries, Ltd., Butterwick Research Laboratories, The Frythe, Welwyn, Herts. (1946.)
Croxall, H. E., B.Sc., Ph.D., N.A.A.S., Elswick Hall, Elswick Park, Newcastle-on-Tyne, 4. (1937.)
Curtis, Miss K. M., M.A., D.Sc., D.I.C., F.L.S., c/o The High Commissioner for New Zealand, 415 The Strand, London. (1917.)
Cury, Dr A., Mycology Laboratory, Instituto Oswaldo Cruz, Caixa Postal 926, Rio de Janeiro, Brazil. (1950.)

Dade, H. A., A.R.C.S., Commonwealth Mycological Institute, Ferry Lane, Kew, Surrey. (1927.)

Damiano, C., 81 Exeter House, Putney Heath, London, S.W. 15. (1946.)

Darlington, R. G., Southcourt Road, Worthing, Sussex. (1949.)

Davies, D. L. G., Ph.D., N.A.A.S. Crosswood, Aberystwyth, Cards. (1938.)

Davies, D. W., B.Sc., N.A.A.S., Quebec, Llanbadarn Road, Aberystwyth. Cards. (1923.)

Davies, H. M., B.Sc., Ph.D., 43 Garrick Close, Walton-on-Thames, Surrey. (1944.)

Davis, Mrs L. H., Division of Plant Pathology, University of California, Agricultural Experiment Station, Los Angeles, 24, California, U.S.A. (1949.)

Day, W. R., B.A., B.Sc., Imperial Forestry Institute, Oxford. (1928.)

Deighton, F. C., M.A., O.B.E., Mycologist, Department of Agriculture, Njalo via Mano, Sierra Leone, West Africa. (1925.)

Dennis, R. W. G., Ph.D., The Herbarium, Royal Botanic Gardens, Kew, Surrey. (1932.)

De Silva, R., Division of Medical Mycology, Medical Research Institute, Colombo, Ceylon. (1951.)

Dickinson, S., Ph.D., School of Agriculture, Cambridge. (1921.)

Digilio, Dr A. P. L., Instituto Miguel Lillo, Calle Miguel Lillo 205, Tucumán, Argentina. (1952.)

Dobbs, C. G., B.Sc., Ph.D., University College of North Wales, Department of Botany, Memorial Buildings, Bangor. (1933.)

Dodge, Dr C. W., Missouri Botanical Garden, 2315 Tower Grove Avenue, St Louis, 10, Missouri, U.S.A. (1926.)

D'Oliviera, Professor B., Departamento de Fitopatologia, Estação Agronomica Nacional, Sacavem, Portugal. (1939.)

Dorey, M. J., New Timbers, Laleham-on-Thames, Staines, Middx. (1950.)

Dovaston, H. F., West of Scotland Agricultural College, Auchincruive, Ayr. (1946.)

Duddington, C. L., M.A., F.L.S., Ph.D., Biology Department, The Polytechnic, 309 Regent Street, London, W. 1. (1946.)

Dulaney, Dr E. L., Research and Development Division, Merck & Co., Inc., Rahway, New Jersey, U.S.A. (1950.)

Duncan, Miss C. W. H., Glaxo Laboratories Ltd., Ulverston, Lancs. (1949.)

Duncan J. T., F.R.C.S., L.R.C.P. (Ire), 31 Nightingale Road, Rickmansworth, Herts. (1930.)

Dunston, Capt. A. E. A., Poulahash, Ballycorus Road, Kilternan, Co. Dublin, Eire. (1937.)

Dunston, T. F. G. W., Burltons, Donhead St Mary, nr. Shaftesbury, Dorset. (1947.)

Eades, B. W., 83 Selwyn Road, Edgbaston, Birmingham, 16. (1952.)

Ebben, Miss M. H., Experimental & Research Station, Cheshunt, Herts. (1951.)

Edwards, R. L., Ph.D., B.Sc., F.R.I.C., Mushroom Research Association Ltd., Yaxley, Peterborough, Northants. (1951.)

Elliott, A. L., 29 High Street, Pershore, Worcs. (1949.)

Elliott, Mrs J. S. B., D.Sc., Dulverton, Manor Way, Aldwick Bay, nr. Bognor Regis, Sussex. (1911.)

Ellis, E. A., F.L.S., Castle Museum, Norwich. (1937.)

Ellis, M. B., B.Sc., Ph.D., Commonwealth Mycological Institute, Ferry Lane, Kew, Surrey. (1945.)

Emerson, R., Department of Botany, University of California, Berkeley, California, U.S.A. (1938.)

English, Miss M. P., M.Sc., Physiological Department, B. D. H. Ltd., Graham St., City Road, London, N. 1. (1943.)

Esplan, Mrs C., The Cottage, King's Arms Hill, Arundel, Sussex. (1949.)

Evans, B. D., B.Sc., Pontycelyn, 29 Lady Mary Road, Cardiff. (1951.)

Evans, E., Botany School, Downing Street, Cambridge. (1952.)

Ferns, J. F., M.Sc., A.I.C., 16 Lawn Crescent, Kew, Surrey. (1942.)

Fincher, F., Randan Wood, Woodcote, Bromsgrove, Worcs. (1948.)
Findlay, W. P. K., D.Sc., A.R.C.S., Forest Products Research Laboratory, Princes Risborough, Aylesbury, Bucks. (1928.)
Fleetwood, Miss P. M., B.Sc., Department of Botany, The University, Edgbaston, Birmingham, 15. (1951.)
Flentje, N. T., B.Agr.Sci., M.Sc., c/o Waite Institute, Private Mail Bag, Adelaide, S. Australia. (1949.)
Fletcher, W. W., B.Sc., Department of Botany, West of Scotland Agricultural College, Blythswood Square, Glasgow, C. 2. (1948.)
Foister, C. E., B.A., Ph.D., Plant Pathology Service, Seed Testing Station, East Craigs, Corstorphine, Edinburgh, 12. (1940.)
Fothergill, P. G., B.Sc., Ph.D., King's College, Newcastle-on-Tyne, 2. (1946.)
Fox, R. A., B.Sc., B.Agr., Department of Botany, Queen's University, Belfast, N. Ireland. (1951.)
Frank, Mrs E. M., Box 31, Oakglen Station, Lansing, Illinois, U.S.A. (1943.)
Fraser, Miss L. R., D.Sc., Biological Branch, Department of Agriculture, Box 36A, G.P.O., Sydney, Australia. (1938.)
Fraymouth, Miss. J. Department of Botany, The University, Bristol. (1950.)
Fries, Dr N., Institute of Physiological Botany, The University, Uppsala, Sweden. (1946.)
Fulford, R. M., Department of Botany, University of Nottingham. (1949.)
Fulton, D. D. C., The Laurels, Hadnall, nr. Shrewsbury, Salop. (1952.)
Galleymore, Miss H. B., Pest Infestation Laboratory, London Road, Slough, Bucks. (1948.)
Garrett, S. D., M.A., D.I.C., Sc.D., Botany School, Downing Street, Cambridge. (1936.)
Gates, L. F., B.A., Rothamsted Experimental Station, Dunholme Field Station, Dunholme, Lincoln. (1947.)
Gay, J. L., B.Sc., M.Sc., School House, Hartpury, Gloucester. (1952.)
Gaze, R. W. T., Pathological Laboratory, Royal Infirmary, Leics. (1949.)
Geddes, J. A., Department of Botany, University of Oxford, South Parks Road, Oxford. (1951.)
Gemmell, Professor A. R., B.Sc., Ph.D., M.S., Department of Biology, University College of North Staffordshire, Keele, Stoke-on-Trent, Staffs. (1946.)
Gentles, J. C., B.Sc., Department of Botany, University of Glasgow. (1948.)
Gibbon, Miss O. M., B.Sc., Red Rice, Timsway, Chertsey Lane, Staines, Middx. (1952.)
Gibson, I. A. S., M.A., The Forest Department, P.O. Box 337, Nairobi, Kenya. (1952.)
Gilbert, E. J., Docteur en Pharmacie, 4 Rue de Musset, Paris (16^{e}), France. (1924.)
Gillespie, Dr J., Country Gardens, Rainford End, Chelmsford, Essex. (1938.)
Glasscock, H. H., Ph.D., Wye College, Wye, Ashford, Kent. (1949).
Glen-Bott, Miss J. I., Bunchwood, Bunch Lane, Haslemere, Surrey. (1949.)
Glynn, E. St. A., 6A Lancaster Drive, London, N.W. 3. (1946.)
Glynne, Miss M. D., D.Sc., Rothamsted Experimental Station, Harpenden, Herts. (1932.)
Gobie, C. H., B.Sc., F.R.G.S., 4 Melton Court, Amyand Park Road, Twickenham, Middx. (1953.)
Goldie-Smith, Miss E. K., Department of Botany, University of North Carolina, U.S.A. (1944.)
Goodchild, N. A., B.Sc.Agric., P.O. Box 91, Kericho, Kenya. (1951.)
Goodman, Miss C. R., 2 Victoria Road, Harborne, Birmingham, 17. (1951.)
Goodman, R. E. J., Norfolk Flax Establishment, Flitcham Abbey, Flitcham, King's Lynn, Norfolk. (1944.)
Graddon, W. D., B.Sc., A.M.I.Chem.E., 'Rathgar', Park Lane, Congleton, Cheshire. (1942.)

Grainger, Miss J., Wilshaw Road, Meltham, nr. Huddersfield. (1943.)

Grainger, J., B.Sc., Ph.D., Department of Plant Pathology, West of Scotland Agricultural College, Auchincruive, Ayr. (1944.)

Gray, Miss E. G., B.Sc., Ph.D., North of Scotland Agricultural College, 41½ Union Street, Aberdeen. (1942.)

Gregg, Miss M., B.Sc., Ph.D., 12 Bushey Park Road, Rathgar, Dublin, Eire. (1948.)

Gregory, P. H., Ph.D., D.Sc., F.L.S., Rothamsted Experimental Station, Harpenden, Herts. (1930.)

Griffiths, D. J., M.Sc., Welsh Plant Breeding Station, Aberystwyth. (1951.)

Griffiths, E. G., Agricultural Buildings, Alexandra Road, Aberystwyth. (1948.)

Groves, Dr J. W., Division of Botany and Plant Pathology, Science Service Building, Ottawa, Ontario, Canada. (1942.)

Grossbard, Miss E., Department of Botany, Imperial College of Science and Technology, South Kensington, London, S.W. 7. (1948.)

Gupta, S. C., M.Sc., Department of Botany, Imperial College of Science and Technology, South Kensington, London, S.W. 7. (1952.)

Gwynne, D. C., N.A.A.S., Plant Pathology Department, Elswick Hall, Elswick Park, Newcastle-on-Tyne, 4. (1946.)

Gwynne-Vaughan, Professor Dame Helen, G.B.E., D.Sc., LL.D., F.L.S., 93 Bedford Court Mansions, London, W.C. 1. (1906.)

Hafiz, A., Ph.D., D.I.C., Ph.D. (Punjab), M.Sc.(Agr.), Punjab Agricultural College and Research Institute, Lyallpur, Pakistan. (1950.)

Hagen, A., Botanisk Museum, Oslo, Norway. (1949.)

Hainsworth, H. E., M.A., Tocklai Experimental Station, Cinnamara, P.O. Assam, India. (1946.)

Hall, Rev. Charles A., F.R.M.S., 'Gables', Sleepy Hollow, nr. Storrington, Sussex. (1948.)

Hams, A. F., B.Sc., A.R.C.S., 37 Swains Avenue, Bakersfields, Nottingham. (1948.)

Hanna, W. F., M.Sc., Ph.D., c/o University of Manitoba, Fort Garry, Manitoba, Canada. (1925.)

Hansford, C. G., M.A., F.L.S., Sc.D. (Cantab.), D.Sc. (Adelaide), University of Adelaide, Waite Agricultural Research Institute, Private Mail Bag, Adelaide, S. Australia. (1921.)

Harley, J. L., M.A., D.Phil., Department of Botany, The University, Oxford. (1932.)

Harris, G. C. M., M.A., D.Phil., c/o Commerical Solvents Corporation, Terrehaut, Indiana, U.S.A. (1946.)

Harris, R. V., D.Sc., A.R.C.S., East Malling Research Station, nr. Maidstone, Kent. (1924.)

Harrison, R. W., B.Sc., 121 Wellsway, Bath, Somers. (1953.)

Harrold, C. E., B.Sc., Ph.D., Department of Botany, The University, Edgbaston, Birmingham, 15. (1951.)

Harthan, Miss B. J., 2 Derbyshire Lane, Stretford, nr. Manchester. (1951.)

Harvey, R., B.Sc., Department of Botany, University College, Newport Road, Cardiff. (1951.)

Haskins, R. H., B.A., M.A., Ph.D., National Research Council, Prairie Regional Laboratory, Saskatoon, Sask., Canada. (1946.)

Hassell, F. C., 8 Sunbury Gardens, Dartry Road, Dublin, S. 2, Eire. (1946.)

Hastings, Somerville, M.S., F.R.C.S., 12 Westminster Palace Gardens, Victoria Street, London, S.W. 1. (1913.)

Hawker, Miss L. E., Ph.D., D.Sc., Botanical Department, The University, Bristol. (1934.)

Hawkins, J. H., B.Sc., M.Sc., Elmwood Cottage, Calverley, nr. Leeds. (1953.)

Heimbeck, Mrs L., Brosoe, Levanger, Norway. (1923.)

Hemming, H. G., Imperial Chemical Industries, Ltd., Butterwick Laboratories, The Frythe, Welwyn, Herts. (1946.)

Henderson, D. M., Royal Botanic Gardens, Inverleith Row, Edinburgh. (1951.)
Herd, G. W., 20 Cefn Carnau Road, The Heath, Cardiff. (1952.)
Herd, Miss S. J., B.Sc., Wellbrae, Bankwell Road, Anstruther, Fife. (1947.)
Hesseltine, C. W., Northern Regional Research Laboratory, Peoria, Ill., U.S.A. (1948.)
Hewett, P. D., 61 Chester Drive, North Harrow, Middlesex. (1947.)
Hewlett, Miss M. A., B.Sc.(Hort.), 3 Upperton Road, Sidcup, Kent. (1953.)
Hickman, C. J., B.Sc., M.Sc., Ph.D., Botanical Department, The University, Edgbaston, Birmingham, 15. (1935.)
Hidaka, Dr Z., Department of Plant Pathology, Hatano Tobacco Experiment Station, Hatano, Kanagawa, Japan. (1951.)
Hilton, R. N., B.A., Rubber Research Institute of Malaya, P.O. Box 150, Kuala Lumpur, Malaya. (1948.)
Hincks, W. D., Manchester Museum, The University, Manchester, 13. (1951.)
Hinson, W. H., B.Sc., Department of Botany, Memorial Buildings, University College of North Wales, Bangor. (1950.)
Hirst, J. M., B.Sc., Rothamsted Experimental Station, Harpenden, Herts. (1949.)
Hiscott, G. S., Furzy Grove, Kempshott Lane, Basingstoke, Hants. (1944.)
Hof, Dr T., Centraal Instituut voor Materiaalonderzoek, Rijswijk (Bij Delft), Lange Kleiweg 22, Holland. (1947.)
Holden Miss M., B.Sc., Rothamsted Experimental Station, Harpenden, Herts. (1950.)
Holliday, P., Imperial College of Tropical Agriculture, St Augustine, Trinidad, British West Indies. (1948.)
Holmes, G. D., University College of North Wales, Bangor, Caern. (1947.)
Hora, Dr F. B., The University, Reading, Berks. (1943.)
Howard, E., B.Sc., Flat 2, Westwood, Reddown Road, Coulsdon, Surrey. (1952.)
Howard, H. J., F.R.M.S., F.L.S., Lingfield, 6 College Road, Norwich. (1918.)
Howe, F. J., F.R.M.S., Pathological Department, St Cross Hospital, Rugby, Warwickshire. (1943.)
Hughes, S. J., M.Sc., Division of Botany and Plant Pathology, Department of Agriculture, Ottawa, Ontario, Canada. (1941.)
Hughes, W. A., East of Scotland College of Agriculture, Edinburgh. (1952.)
Huijsmann, Dr H. S. C., Oogarts, Doetinchem, Holland. (1947.)
Hull, R., B.Sc., Ph.D., A.R.C.S., c/o Rothamsted Experimental Station, Field Research Station, Hackthorn, Lincoln. (1942.)
Humphreys Jones, D. R., B.Sc., Grassland Research Institute, Stratford-on-Avon, Warwicks. (1952.)
Hutchings, G. E., Juniper Hall Field Centre, Dorking, Surrey. (1948.)
Hutchinson, P. B., Imperial College of Tropical Agriculture, Trinidad. (1951.)
Hutchinson, S. A., Ph.D., Department of Botany, The University, Glasgow. (1948.)
Ingold, Professor C. T., D.Sc., Ph.D., F.L.S., Department of Botany, Birkbeck College, Malet Street, London, W.C. 1. (1935.)
Ingram, Miss M. E. K., Lauriston, 9 West Way, Rickmansworth, Herts. (1949.)
Ives, Miss J., 26 Woodlark Road, Cambridge. (1949.)
Jackson, R. M., Department of Agriculture, University College, Ibadan, Nigeria, W. Africa. (1946.)
Jacques-Félix, Madame M., 68 rue François Rolland, Nogent sur Marne (Seine), France. (1952.)
Jarvis, W. R., B.Sc., Plant Pathology Department, Imperial College, South Kensington, London, S.W. 7. (1951.)
Jay, B. A., M.A., F.L.S., 9 Thornton Crescent, Old Coulsdon, Surrey. (1946.)
Jeffreys, E. G., Imperial Chemical Industries Ltd., Butterwick Laboratories, The Frythe, Welwyn, Herts. (1946.)
Jenkins, Miss A. E., Bureau of Plant Industry, Department of Agriculture, Washington, D.C., U.S.A.

Jenkins, J. E. E., Ministry of Agriculture and Fisheries, 33 Turners Hill, Cheshunt, Herts. (1949.)

Johnstone, R. H., 726 Anniesland Road, Glasgow, W. 4. (1938.)

Jones, D. Rudd, M.A., Ph.D., Imperial Chemical Industries Ltd., Butterwick Research Laboratories, The Frythe, Welwyn, Herts. (1949.)

Jones, Miss E. S. L., Syracuse, High Cross Lane, Newport, Mon. (1950.)

Jones, F. G. B., M.A., 32 Kingston Lane, Teddington, Middx. (1949.)

Jørstad, Ivar, Statsmykolog, Botanisk Museum, Oslo, Norway. (1923.)

Juniper, A. J., B.Sc., 50 Dawlish Drive, Ilford, Essex. (1951.)

Justham, Mrs M. C. D., Ph.D., Cherry Dene, Cairn Road, Bielside, Aberdeen. (1945.)

Keay, Miss M. A., M.A., Ph.D., Agricultural Research Council, Plant Genetics Station, Huntingdon Road, Cambridge. (1935.)

Keeping, Dr E. Silver, Provincial Laboratory, University of Alberta, Edmonton, Alberta, Canada. (1950.)

Kennedy, Miss M. E., B.Sc., M.Sc., Hawthorndale Laboratories, Jealott's Hill Research Station, Bracknell, Berks. (1951.)

Kent, W. W., Red House, Rodbourne, Swindon, Wilts. (1948.)

Kerr, A., B.Sc., Waite Agricultural Research Institute, Adelaide, South Australia. (1948.)

Kertland, Miss N. P. H., M.Sc., Department of Botany, Queen's University, Belfast, Northern Ireland. (1948.)

Keys, Mrs V., c/o Dr D. N. Keys, Westminster City Council, 31 Charing Cross Road, London, W.C. 1. (1952.)

Keyworth, W. G., Ph.D., D.I.C., National Vegetable Research Station, Wellesbourne, Warwick. (1941.)

Kharbouch, M. A. M., Department of Botany, The University, Sheffield. (1951.)

Kind, S. S., B.Sc., Boyne Hill Cottage, Chapelthorpe, nr. Wakefield, Yorks. (1952.)

Kingston, B. H., Leogang, 1 Lavender Road, Carshalton, Surrey. (1948.)

Klein, R., Ph.D., F.L.S., 1 Emperor's Gate, London, S.W. 7. (1943.)

Knapman, Miss J., Penn Sylva, Hele Manor, Barnstaple, N. Devon. (1952.)

Knight, B. C., B.Sc., North of Scotland College of Agriculture, 41½ Union Street, Aberdeen. (1951.)

Korf, Professor R. P., Department of Plant Pathology, Cornell University, Ithaca, N.Y., U.S.A. (1947.)

Kramer, L. M. J., M.A., Ph.D., 41 Marlborough Mansions, Cannon Hill, London, N.W. 6. (1946.)

Krebs, Mrs L., 42 Selcroft Road, Purley, Surrey. (1948.)

Langdon, R. F., Department of Botany, University of Queensland, George Street, Brisbane. (1950.)

Lange, M., Universitetets Planteanatomiske Laboratorium, Gothersgade 140, Copenhagen, Denmark. (1949.)

Lapwood, D. H., B.Sc., A.R.C.Sc., 97 Raynton Road, Enfield, Middx. (1935.)

Large, E. C., Ministry of Agriculture, Plant Pathology Laboratory, Milton Road, Harpenden, Herts. (1948.)

Last, F. T., Ph.D., Rothamsted Experimental Station, Harpenden, Herts. (1949.)

La Touche, C. J., M.Sc., L.A.H., Department of Bacteriology, Medical School, University of Leeds. (1946.)

Leach, R., B.A., School of Agriculture, Cambridge. (1929.)

Leather, R. I., B.Sc., Wharfecote, Benrhydding, Ilkley, W. Yorks. (1952.)

Lee, O. M., School of Agriculture, Serdang, Selangor, Malaya. (1946.)

Leech, Mrs E., 12 Alistair Drive, Yeovil, Somerset. (1947.)

Le Gal, Mme Marcel, Laboratoire de Cryptogamie, Muséum National d'Histoire Naturelle, 12 rue de Buffon, Paris. (1948.)

Lelliott, R. A., B.A., 138 Topstreet Way, Harpenden, Herts. (1952.)

Lester, E., N.A.A.S., Shardlow Hall, Shardlow, nr. Derby. (1948.)

Levisohn, Miss J., D.Phil., Bedford College for Women, Regent's Park, London, N.W. 1. (1943.)
Lloyd, Dr Blodwen, Royal Technical College, Glasgow, C 1. (1946.)
Lloyd, Mrs J. W., 63, Charnock Ave., Wollaton Park, Notts. (1949.)
Locquin, M., Muséum National d'Histoire Naturelle, Paris. (1950.)
Lodhi, S. A., M.Sc., Department of Botany, University of the Punjab, Lahore, Pakistan. (1950.)
Lofts, M. J., Nigerian College of Art, Science and Technology, Ibadan, Nigeria. (1947.)
Lohwag, Dr K., Vienna III, Rennweg 2, Austria. (1950.)
Longley, B., Slape Mills, Netherbury, nr. Beaminster, Dorset. (1946.)
Loos, C. A., Tea Research Institute, St Coombs, Talawakele, Ceylon. (1952.)
Lowings, P. H., B.Sc., B.Agr., Mycology of Plant Pathology Dept., Queen's University, Belfast, N. Ireland. (1951.)
Luc, M., Institut Intercolonial d'Adiopodoumé, Boîte Postale 20, Abidjan, Cote d'Ivoire. (1951.)
Lucas, R. L., Department of Agriculture, The University, Oxford. (1949.)
Lupton, F. G. H., M.A., (Agricultural Research Council), Plant Breeding Institute, School of Agriculture, Cambridge. (1952.)
Lütjeharms, Professor W. J., Phil.Nat.D., Department of Botany, University College, Bloemfontein, Orange Free State, S. Africa. (1930.)
Macdonald, J. A., D.Sc. Ph.D., F.R.S.E., Botany Department, The University, St Andrews. (1938.)
McIlwaine, R. S., Plant Pathology Department, Queen's University, Belfast, N. Ireland. (1951.)
McKay, R., B.Sc., A.R.C.S.I., Department of Plant Pathology, Albert Agricultural College, Glasnevin, Dublin, N.W. 3. (1939.)
McKee, R. K., Nottingham University School of Agriculture, Sutton Bonington, Loughborough, Leics. (1947.)
Mackinnon, Juan E., M.D., Instituto de Higiene, Av. A. Ricaldoni 3051, Parque J. Battle y Ordóñez, Montevideo, Uruguay. (1947.)
Madge, Miss M. A. P., Ph.D., Royal Holloway College, Englefield Green, Surrey. (1948.)
Main, Mrs S. M., 74 Jesmond Avenue, Wembley, Middx. (1950.)
Malcolmson, Miss J. F., 25 Coillesdene Drive, Portobello, Midlothian. (1952.)
Malençon, G., Inspecteur Principal, Bureau de la Défense Végétaux, 65 bis Avenue de Témara, Rabat, Morocco. (1946.)
Malone, J. P., B.Sc., Department of Mycology and Plant Pathology, Queen's University, Belfast, N. Ireland. (1948.)
De Man, G., 37 Av. des Mimosas, Woluwé St Pierre, Brussels, Belgium. (1951.)
Mangenot, F., Faculté de Pharmacie, 5 rue de la Prairie, Nancy, M. & M., France. (1953.)
Manners, J. G., B.A., Ph.D., Department of Botany, The University, Southampton. (1948.)
Marcan, Mrs B., 335 West Wycombe Road, High Wycombe, Bucks. (1948.)
Marsh, R. W., M.A., Research Station, Long Ashton, Bristol. (1923.)
Martens, Professor P., 23 Rue Marie Thérèse, Louvain, Belgium. (1947.)
Martin-Scott, I., M.D., 16 Buckingham House, Courtlands, Richmond, Surrey. (1948.)
Mason, E. W., M.A., M.Sc., F.L.S., Commonwealth Mycological Institute, Ferry Lane, Kew, Surrey. (1921.)
Mason, Mrs E. W., M.Sc., Inglenook, 63 King's Road, Richmond, Surrey. (1922.)
Mathias, W. T., B.Sc., Department of Botany, The University, Liverpool. (1938.)
Mathieson, Miss M. J., M.Sc., Botany Department, University of Melbourne, Australia. (1947.)
Maxted, Mrs B., B.Sc., Ph.D., Department of Botany, The University, Reading. (1934.)
Mayers, T. T., Queensway Hotel, 11 Prince's Square, London, W. 2. (1949.)

Melin, Professor J. B. E., Institute of Physiological Botany, Uppsala, Sweden. (1946.)

Mence, Miss M. J., School of Agriculture, University College of North Wales, Bangor, Caern. (1948.)

Menzies, Miss B. P., Department of Botany, Bedford College, Regent's Park, London, N.W. 1. (1948.)

Miller, Professor J. H., B.S., M.S., Ph.D., University of Georgia, Athens, Ga., U.S.A. (1930.)

Montgomerie, Miss I. G., Seed Testing, Plant Registration and Plant Pathology Station, East Craigs, Corstorphine, Edinburgh, 12. (1951.)

Montgomery, H. B. S., B.A., Ph.D., D.I.C., Research Station, East Malling, nr. Maidstone, Kent. (1940.)

Montgomery, Mrs N., 'Chelmsley', Tollerton Lane, Tollerton, Notts. (1949.)

Moon, J. M^cK., Finglush, Station Road, Greenisland, Co. Antrim, N. Ireland. (1948.)

Moore, Miss F. J., Ph.D., Ministry of Agriculture, Plant Pathology Laboratory, Milton Road, Harpenden, Herts. (1945.)

Moore, M. H., M.Sc., Ph.D., Research Station, East Malling, nr. Maidstone, Kent. (1940.)

Morquer, Professor R., Faculté des Sciences, Laboratoire de Botanique Générale, Toulouse, France. (1951.)

Morris, L. E., c/o Eton College, Windsor, Berks. (1924.)

Morris, P. E., B.Sc., D.I.C., M.R.I.P.H.H., Fernside, Ormesby, St. Margaret, Norfolk. (1950.)

Moser, M., Innsbruck, Mullerstrasse 9, Austria. (1949.)

Mount, Miss S., Department of Botany, Imperial College of Science and Technology, South Kensington, London, S.W. 7. (1948.)

Moureau, Dr J., Chef du Centre de Recherches Scientifiques, Irsac, Bikoro, Coquilhatville, Belgian Congo. (1944.)

Munro, H. R., The Uplands, Monument Lane, Rednal, Birmingham. (1948.)

Munson, R. G., 29 Northwick Circle, Kenton, Harrow, Middlesex. (1943.)

Murray, J. S., Forest Research Station, Alice Holt Lodge, nr. Wrecclesham, Farnham, Surrey. (1950.)

Muskett, Professor A. E., D.Sc., A.R.C.S., M.R.I.A., Department of Mycology and Plant Pathology, Queen's University, Belfast, N. Ireland. (1923.)

Nannfeldt, Professor J. A., Institute of Systematic Botany, University of Uppsala, Uppsala 1, Sweden. (1932.)

Nash-Wortham, J. R. H., 9 Harrington Road, Preston Park, Brighton, Sussex. (1937.)

Nattrass, R. M., B.Sc.(Agric.), Ph.D., Department of Agriculture, P.O. Box 338, Nairobi, Kenya. (1925.)

Nelson, J. D. E., B.Sc., Glaxo Laboratories Ltd., North Lonsdale Road, Ulverston, Lancs. (1952.)

Newby, H. V., Department of Botany, Queen Mary College, London, E. 1. (1946.)

Newhook, F. J., M.Sc., Ph.D., D.I.C., Plant Diseases Division, D.S.I.R., Private Bag, Auckland, New Zealand. (1946.)

Nicholls, Miss V. O., Department of Botany, University of London, King's College, 68 Half Moon Lane, Herne Hill, S.E. 24. (1949.)

Nobbs, Miss G. E. D., 4 Clifton Estate, Plympton, S. Devon. (1943.)

Noble, Miss M., B.Sc., Ph.D., 19A Willowbrae Avenue, Edinburgh. (1940.)

Norman, Miss T., B.Sc., 'Chiltern Court', St Peter's Avenue, Caversham, Reading, Berks. (1953.)

Nour, Mohammed, A., B.Sc., School of Agriculture, Shambat, Sudan. (1951.)

O'Byrne, J. K., 15 Parklands Court, Hounslow West, Middx. (1950.)

O'Connor, P., Ph.D., B.Sc., A.R.C.Sc.I., National Museum, Dublin, Eire. (1925.)

Ogilvie, L., M.A., M.Sc., N.A.A.S., Bracken Hill, Leigh Woods, Bristol 8. (1922.)

Olivier, Miss D. L., Department of Botany, Rondebosch University, Cape Province, S. Africa. (1950.)
Orton, P. D., M.A., A.R.C.M., Epsom College, Epsom, Surrey. (1947.)
Owen, H., B.S.c., A.I.C.T.A., Department of Agricultural Botany, The University, Reading. (1950.)
Padwick, Dr G. W., Imperial Chemical Industries, Ltd., Nobel House, 2 Buckingham Gate, London, S.W. 1. (1936.)
Page, Miss W. M., M.Sc., Ph.D., 5 Dartmouth Chambers, Theobald's Road, London, W.C. 1. (1921.)
Palmer, J. T., 5 Station Road, Woodley, Stockport, Cheshire. (1950.)
Parker, H. M., Brereton, Lyth Hill, Road, Bayston Hill, Shrewsbury, Salop. (1949.)
Parker-Rhodes, A. F., 20 Sedley Taylor Road, Cambridge. (1950.)
Parkinson, D., Department of Botany, The University, Nottingham. (1950.)
Parkinson, Miss S. E., B.Sc., 46 St Marks Crescent, Maidenhead, Berks. (1952.)
Paton, Miss M. R., Barnbarroch House, Dalbeattie, Scotland. (1945.)
Peach, Mrs M., 5 Tybenham Road, Merton Park, London, S.W. 19. (1947.)
Pegg, G. F., B.Sc., 28 Vale Street, Derby. (1952.)
Peklo, Professor Dr J., Institute of Phytopathology, College of Agriculture and Forestry, Prague XIX, Dejvice, Czechoslovakia. (1924.)
Peters, J. H., 64 Salisbury Street, Bedford. (1953.)
Pettifer, A. J., 36 Cheviot Gardens, Hendon Way, London, N.W. 2. (1944.)
Peyronel, Professor B., Directeur, Instituto & Orto Botanico, Viale Mattioli 25 (Valentino), Torino, Italy. (1932.)
Phillips, D. H., M.Sc., The State Experimental Station, Howard Davis Farm, Trinity, Jersey, Channel Islands. (1950.)
Phillips, Dr H. H., 11 King's End, Bicester, Oxon. (1923.)
Piane, Monsieur V., Martignat (Ain), France. (1952.)
Piggott, Dr C. S., Science Office, American Embassy, 1 Grosvenor Square, London. (1953.)
Pilát, Dr A., Department of Botany, National Museum, Prague II, Václavské nám. 1700, Czechoslovakia. (1946.)
Ping, A. Wentworth, M.A., "Lowfields", 8 Galtres Rd., Clifton Without, York. (1926.)
Pinto-Lopes, Dr J., Faculdade de Ciências, Lisbon, Portugal. (1949.)
Platt, J., B.Sc., 'The Chestnuts', St Andrew's Road, Burnham-on-Sea, Somerset. (1947.)
Plunkett, B. E., B.Sc., Ph.D., Department of Botany, Birkbeck College, Malet Street, London, W.C. 1. (1945.)
Pollard, H. B. C., 13 Abercorn Place, London, N.W. 8. (1949.)
Porter, Mrs L., M.Sc., Summerfield, Dalkey Avenue, Dalkey, Co. Dublin, Eire. (1949.)
Porter, S. C., A.R.I.C., A.M.I.Chem.E., 57 Billesley Lane, Moseley, Birmingham, 13. (1946.)
Prentice, I. W., B.Sc., Ph.D., Ministry of Agriculture, Plant Pathology Laboratory, Milton Road, Harpenden, Herts. (1943.)
Preston, N. C., B.Sc., N.A.A.S., Woodthorne, Wolverhampton, Staffs. (1920.)
Price, E. A. S., B.A., Cambridge Cottage, Low Ackworth, nr. Pontefract, Yorks. (1953.)
Price, J. W., The Moor, Eardiston, nr. Tenbury, Worcs. (1952.)
Priest, D., 308 Hither Green Lane, Lewisham, London, S.E. 13. (1952.)
Prowse, G. A., c/o Department of Botany, University College, Khartoum, Sudan. (1949.)
Pugh, G. J. F., Department of Botany, University of Nottingham. (1952.)
Quick, A. H., B.Sc., A.R.I.C., Greengarth, Quarryside Road, Mirfield, Yorks. (1952.)
Ralph, P. H. A., 6A Ellenslea Road, St Leonards-on-Sea. (1950.)
Rapson, Mrs H., 307 Grenville House, Dolphin Square, London, S.W. 1. (1950.)
Rautavaara, Dr T., Mannerheimint 56E 37, Helsinki, Finland. (1949.)
Ray, Miss A., Penarwyn, Gorran Haven, Gorran, Cornwall. (1929.)
Rayner, R. W., The Coffee Research Station, Box 4, Ruiru, Kenya, E. Africa. (1925.)

Rees, J., M.Sc., Y Faerdref, 404 Cyncoed Road, Cardiff. (1929.)

Reid, D. A., 19 High Street, Leighton Buzzard, Beds. (1950.)

Richards, M., Ph.D., Asthma and Allergy Research Unit, St David's Hospital, Cardiff. (1950.)

Riddell, R. W., M.D., M.R.C.P., Department of Medical Mycology, Institute of Dermatology, St John's Hospital for Diseases of the Skin, Lisle Street, Leicester Square, London, W.C. 2. (1950.)

Ritson, W., 12 West Street, Winwick Road, Warrington, Lancs. (1946.)

Roberts, E. T., B.Sc., A.R.C.S., N.A.A.S., 'Chiltern Court,' St Peter's Avenue, Caversham, Reading, Berks. (1947.)

Roberts, Miss F. M., Ph.D., Rothamsted Experimental Station, Harpenden, Herts. (1948.)

Roberts, Miss M., 7 Manson Place, London, S.W. 7. (1949.)

Robertson, A. G., B.Sc., 2 Westfield Park, Redland, Bristol 6. (1948.)

Robertson, N. F., Ph.D., Botany School, Downing Street, Cambridge. (1945.)

Robinson, E., 26 Burwood Avenue, Eastcote, Pinner, Middx. (1938.)

Rogers, Dr D. P., New York Botanical Garden, Bronx Park, New York 58, N.Y., U.S.A. (1943.)

Rogerson, C. T., Department of Botany and Plant Pathology, Kansas State College, Manhattan, Kansas, U.S.A. (1947.)

Roscoe-Abbott, Capt. J., B.Sc., The School House, Withern, Alford, Lincs. (1946.)

Rose, H. V., Morden College, Blackheath, London, S.E. 3. (1944.)

Rosser, W. R., 15 Knox Road, Blakenhall, Wolverhampton. (1950.)

Rousset-Hall, O., M.A., 44 Great Bowden Road, Market Harborough, Leics. (1942.)

Roy, R. Y., Benares Hindu University, India. (1947.)

Saich, Miss P. M., B.Sc., Department of Botany, The University, Edgbaston, Birmingham, 15. (1951.)

Salt, G. A., M.A., Plant Pathology Department, Rothamsted Experimental Station, Harpenden, Herts. (1946.)

Sampson, Miss K., M.Sc., Malmsmead, Lacey Green, nr. Aylesbury, Bucks. (1920.)

Samuel, G., M.Sc., Agricultural Research Council, Cunard Building, 15 Regent Street, London, S.W. 1. (1923.)

Sansome, Mrs E. R., M.Sc., Botanical Deaprtment, The University, Ibadan, Nigeria. (1945.)

Sarazin, A., 142 Boulevard Maurice Berteaux, Carrières-sur-Seine, S. et O., France (1951.)

Sarkar, Miss S., B.A.

Sattar, A., Ph.D., D.I.C., Assoc. I.A.R.I., B.Sc.(Ag.), Punjab Agricultural College and Research Institute, Lyallpur, Pakistan. (1950.)

Savory, J. G., B.Sc., Forest Products Research Laboratory, Princes Risborough, nr. Aylesbury, Bucks. (1946.)

Schärer-Bider, W., Peter Rot-Str. 58, Bâle, Switzerland. (1947.)

Schimmer, Miss F. C., M.Sc., Wye College, Wye, Ashford, Kent. (1943.)

Scott, M. R., B.Sc., Department of Botany, The University, Edgbaston, Birmingham, 15. (1951.)

Sewell, G. W., B.Sc., 69 Berrylands Road, Surbiton, Surrey. (1952.)

Shaffer, R. L. B. S., M.S., Department of Plant Pathology, Cornell University, Ithaca, New York. (1953.)

Shear, Dr C. L., 1707 North Sixth Street, Monroe, Louisiana, U.S.A. (1930.)

Sheard, Mrs E., Fairfield Farm, Esprick, nr. Kirkham, Lancs. (1937.)

Shepherd, Miss A. M., 50 Riverside Drive, Mitcham, Surrey. (1951.)

Sirry, A. R., B.Sc., Department of Plant Pathology, West of Scotland Agricultural College, Auchincruive, Ayr. (1950.)

Sisley, C., Cuprinol Department, Robbialac Paints, 36 St James's Street, London, S.W. 1. (1947.)

Skene, Professor M., Department of Botany, The University, Bristol, 7. (1936.)

Smith, C. E. D., Imperial Chemical Industries, Ltd. Hawthorndale Laboratories, Jealott's Hill Research Station, Bracknell, Berks. (1949.)

Smith, G., M.Sc., F.R.I.C., London School of Hygiene and Tropical Medicine, Keppel Street, London, W.C. 1. (1936.)

Smith, H. C., Emmanuel College, Cambridge. (1951.)

Smith, N. J. G., Ph.D., B.Sc., c/o Barclay's Bank, 1 Cockspur Street, London, S.W. 1. (1924.)

Snow, D., Ph.D., Department of Agricultural Botany, The University, Reading. (1947.)

Sobels, Miss J. C., Bach.Mast.Sc., Ph.D., Botanisch Laboratorium, Lange Nieuwstraat 106, Utrecht, Holland. (1953.)

Sowter, F. A., F.L.S., 9 North Avenue, Leicester. (1942.)

Sparrow, Dr F. K., Department of Botany, University of Michigan, Ann Arbor, Mich., U.S.A. (1946.)

Spence, J., B.Sc., 10A Ellerslie Park, St Clair, Port of Spain, Trinidad, B.W. Indies. (1951.)

Spilsbury, J. F., B.Sc., Wellcome Research Laboratories, Langley Court, Beckenham, Kent. (1946.)

Stakman, Professor E. C., University of Minnesota, Department of Agriculture, University Farm, St Paul 8, Minn., U.S.A. (1922.)

Stamps, Miss D. J., B.Sc., Ph.D., Commonwealth Mycological Institute, Ferry Lane, Kew, Surrey. (1948.)

Stenton, H., Ph.D., University College, Hull. (1948.)

Stephens, Miss E. L., B.A., Department of Botany, University of Cape Town, South Africa. (1928.)

Stevenson, Dr J. A., 4113 Emery Place, N.W. Washington 16, D.C., U.S.A. (1927.)

Stewart, Miss D. D., 21 Keats Grove, Hampstead, London, N.W. 3. (1946.)

Steyaert, R. L., Ing.A.I.G., 159 Rue des Atrébates, Etterbeek, Brussels IV, Belgium. (1931.)

Stirrup, H. H., M.Sc., N.A.A.S., Shardlow Hall, Shardlow, nr. Derby. (1922.)

Stockdale, Miss P. M., B.Sc., Department of Botany, University College of the South West, Hatherly Laboratories, Prince of Wales Road, Exeter. (1950.)

Stone, B. A., c/o Senior Representative, Department of Supply, 87 Jermyn Street, London, S.W. 1. (1952.)

Storey, H. H., M.A., Ph.D., C.I.M.G., F.R.S., East Africa Agriculture and Forestry Organization, P.O. Box 21, Kikuyu, Kenya. (1922.)

Storey, I. F., Ph.D., N.A.A.S., Anstey Hall, Trumpington, Cambridge. (1941.)

Stubbs, J., B.Sc., Ph.D., Imperial Chemical Industries Ltd., Hawthorndale Laboratories, Jealott's Hill Research Station, Bracknell, Berks. (1947.)

Sullivan, J. B., 5 Dudley Road, Hastings, Sussex. (1951.)

Sutton, W. D., 313 Wharncliffe Road, N., London, Ontario, Canada. (1953.)

Talbot, P. H. B., Ph.D., P.O. Box 994, Pretoria, South Africa. (1945.)

Talboys, P. W., Plant Pathology Department, Research Station, East Malling, Kent. (1948.)

Taylor, J. C., B.Sc., B.Agr., Department of Mycology and Plant Pathology, Queen's University, Belfast, N. Ireland. (1948.)

Taylor, J. W. H., 4 Leybourne Road, Long Lane, Hillingdon, Middx. (1951.)

Terry, H. J., B.Sc.(Hort.), c/o Messrs May & Baker, Ltd., Horticultural Research Station, Boarded Barns Farm, Ongar, Essex. (1953.)

Thomas, D. G., Heathfield, Upper Tumble, Llanelly, Carmarthen. (1946.)

Thomas, Miss E. M., Department of Botany, The University, Edgbaston, Birmingham, 15. (1949.)

Thomas-Perrott, Mrs P. E., Education Department, University College of North Staffordshire, Keele, Staffs. (1945.)

Thompson, Miss J., B.Sc., Ph.D., Department of Biology, College of Technology and Commerce, Leicester. (1947.)

Thornton, R. H., M.Agr.Sc., Ph.D., Soil Bureau, Department of Scientific and Industrial Research, Wellington, New Zealand. (1952.)

Thorold, C. A., M.A., A.I.C.T.A., M.I.Biol., Strathculm, Hele, Exeter, Devon. (1952.)

Thorpe, Dr I. G., 11 Butt's Hill Road, Woodley, Berkshire. (1951.)

Thurston, Miss J. M., B.Sc., Rothamsted Experimental Station, Harpenden, Herts. (1942.)

Tiley, G. E. D., Department of Botany, The University, Bristol 8. (1951.)

Tipler, R. V., B.Sc., A.R.C.S., Imperial Chemical Industries Ltd., Hawthorndale Laboratories, Jealott's Hill Research Station, Bracknell, Berks. (1945.)

Tomkins, R. G., M.A., Ph.D., Director, Ditton Laboratory, East Malling, nr. Maidstone, Kent. (1925.)

Tomlinson, J. A., B.Sc., Ph.D., National Vegetable Research Station, Wellsbourne, Warwick. (1948.)

Topping, Mrs M. D., B.Sc., 40 Walpole Street, Derby. (1933.)

Topping, Mrs M. P., Angroban, Fir Tree Road, Leatherhead, Surrey. (1947.)

Tribe, H. T., School of Agriculture, Cambridge. (1949.)

Trouern-Trend, K., M.Sc., A.R.C.Sc., D.I.C., 11 Hilltop Way, Paul's Dene, Salisbury, Wilts. (1946.)

Tuley, P., Christ's College, Cambridge. (1951.)

Tunstall, A. C., 45 Livingstone Road, Handsworth, Birmingham, 20. (1933.)

Turner, Miss D. M., M.Sc., Department of Agriculture, The University, Leeds. (1950.)

Turner, Miss E. M., B.A., Ph.D., Old School House, Great Haseley, Oxon. (1940.)

Turner, Mrs F. M., Rosecot, 3 Clevedon Road, Lower Tilehurst, Reading, Berks. (1943.)

Turner, Mrs M., M.Sc., Department of Botany, The University, Notts. (1945.)

Twyman, E. S., B.Sc., M.Sc., Ph.D., Schonland Botanical Laboratory, Rhodes University, Grahamstown, S. Africa. (1942.)

Usher, G., 27 Park Wern Road, Sketty, Swansea, Glam. (1951.)

van Beverwijk, Miss A. L., Eemnesserweg 42, Baarn, Holland. (1947.)

Vanterpool, T. C., M.Sc., Department of Botany, University of Saskatchewan, Saskatoon, Canada. (1929.)

Vaughan, Miss R. K. G., Swinton House, Norwich Road, Cromer, Norfolk. (1950.)

Venkatarayan, S. V., Mycologist, Agricultural Department, Bangalore, S. India. (1935.)

Verma, Dr G. S. Botany Department, The University, Lucknow. (1951.)

Viennot-Bourgin, Professor G., Institut National Agronomique, 16 Rue Claude Bernard, Paris 6^{e}. (1936.)

Vincent, Miss M., Paddington Baths, London, W. 10. (1949.)

Wadham, Professor S. M., M.A., Department of Agriculture, The University, Melbourne, Victoria, Australia. (1922.)

Waldie, J. S. L., B.Sc., C.D.A., Department of Agricultural Botany, The University, Reading. (1925.)

Walker, Mrs J., Ph.D., London School of Hygiene and Tropical Medicine, Keppel Street, London, W.C. 1. (1947.)

Wallace, E. R., M.A., N.A.A.S., Shardlow Hall, Shardlow, nr. Derby. (1934.)

Wallace, G. B., B.Sc.(Agric.), Ph.D., Lyamungu, Moshi, Tanganyika Territory, E. Africa. (1928.)

Wallace, T. J., Goodmans, Membury, nr. Axminster, Devon. (1951.)

Walne, Mrs P. R., B.A., Bryn Siriol, St Agnes Road, Conway, Caerns., N. Wales. (1949.)

Walters, N. E. M., B.Sc., Plant Industry Division, C.S.I.R.O., P.O. Box 109, City, Canberra, A.C.T., Australia. (1952.)

Warcup, J. H., M.Sc., Ph.D., Waite Agricultural Research Institute, Private Mail Bag, Adelaide, South Australia. (1947.)

Ward, Miss G. M., Ph.D., 8 Richmond Road, Cambridge. (1950.)

Wardlaw, Professor C. W., Department of Cryptogamic Botany, The University, Manchester, 13. (1943.)
Warne, Mrs M. M., B.A., 25 Amherst Road, Fallowfield, Manchester, 14. (1937.)
Warren, Dr Clara M., M.R.C.S., L.R.C.P., Fiveways, Leybourne Park, Kew, Richmond, Surrey. (1946.)
Waterhouse, Miss G. M., M.Sc., Commonwealth Mycological Institute, 18 Cumberland Road, Kew, Surrey. (1927.)
Waterston, J. M., B.Sc., Ph.D. (1934.)
Watson-Walker, J., 12 The Bowley, Diseworth, Derby. (1949.)
Webb, P., Ph.D., Clare Cottage, East Malling, nr. Maidstone, Kent. (1938.)
Webb, R. A., B.A., D.Phil., National Hospital, Queen's Square, London, W.C. 1. (1936.)
Webster, B. N., Tea Research Institute of Ceylon, St Coombs, Talawakelle, Ceylon. (1948.)
Webster, J., B.Sc., Department of Botany, The University, Sheffield, 10. (1945.)
Westerdijk, Professor Johanna, Javalaan 4, Baarn, Holland. (1923.)
Western, J. H., B.Sc., Ph.D., Department of Agriculture, The University, Leeds, 2. (1934.)
Weston, W. A. R. D., M.A., Ph.D., N.A.A.S., Anstey Hall, Trumpington, Cambridge. (1923.)
White, N. H., D.Sc., Faculty of Agriculture, The University, Sydney, New South Wales, Australia. (1944.)
Whitehead, T., D.Sc., A.R.C.S., N.A.A.S., Bryn Adda, Bangor, Caern. (1920.)
Whitehouse, H. L. K., Botany School, Downing Street, Cambridge. (1948.)
Wilkins, W. C. (1947.)
Wilkins, W. H., M.A., D.Phil., St Peter's Hall, Oxford. (1928.)
Wilkinson, E. H., B.Sc., Ph.D., Wye College, Wye, Ashford, Kent. (1938.)
Willetts, H. J., 38 Hillside Terrace, Bedwas, Mon. (1950.)
Williams, P. H., B.Sc., Experimental and Research Station, Cheshunt, Herts. (1930.)
Wilson, Miss A. P., M.B.E., A.R.C.S., 7A Arlington Drive, Nottingham. (1929.)
Wilson, A. R., Ph.D., Nottingham University School of Agriculture, Sutton Bonington, Loughborough. (1933.)
Wilson, Miss I. M., B.Sc., Ph.D., Department of Botany, University College of Wales, Aberystwyth. (1938.)
Wilson, J. F., B.Sc., Juniper Cottage, Nettlestead, nr. Maidstone, Kent. (1950.)
Wilson, M., D.Sc., A.R.C.S., F.L.S., Royal Botanic Garden, Edinburgh. (1912.)
Wiltshire, S. P., D.Sc., Commonwealth Mycological Institute, Ferry Lane, Kew, Surrey. (1920.)
Witts, K. J., B.Sc., Messrs A. Guinness, Son & Co., Ltd., Ockham, Ewhurst, Robertsbridge, Sussex. (1952.)
Wolf, Dr F. A., Department of Botany, Duke University, Durham, North Carolina, U.S.A. (1940.)
Wolf, Dr F. T., Vanderbilt University, Nashville, Tennessee, U.S.A. (1940.)
Wood, F. C., The Rest, Franklin Road, Durrington, Worthing. (1935.)
Wood, R. K. S., Ph.D., Department of Botany, Imperial College of Science and Technology, South Kensington, London, S.W. 7. (1946.)
Wood-Baker, Mrs A. W., Birkbeck College, Malet Street, London, W.C. 1. (1948.)
Woodcock, A. J. A., M.Sc., F.E.S., Rhianva, 65 Rock Avenue, Gillingham, Kent. (1926.)
Woodward, R. C., Ph.D., Overseas Development Dept., Plant Protection Ltd., Bolton House, 61 Curzon Street, Piccadilly, London, W. 1. (1924.)
Wormald, H., D.Sc., A.R.C.S., 66 Bower Mount Road, Maidstone, Kent. (1921.)
Wright, B. G., B.Sc., Imperial Chemical Industries, Ltd., Dyestuffs Division, Trafford Park, Manchester, 17. (1950.)

Wright, Dr F. R. E., Braunton, N. Devon. (1946.)
Wright, Miss J. M., Imperial Chemical Industries, Ltd., Butterwick Research Laboratories, The Frythe, Welwyn, Herts. (1949.)
Wyatt, F. M. (1949.)
Yuill, E., 35 Askham Lane, Acomb, York. (1937.)

Associate Members

Bolton, Miss M., Oakleigh, 20 Druid Road, Stoke Bishop, Bristol 9. (1947.)
Bray, Miss M. F., Imperial Chemical Industries, Ltd., Hawthorndale Laboratories, Jealott's Hill Research Station, Bracknell, Berks. (1949.)
Brokenshire, F. A., 2 Rock Avenue, Barnstaple, N. Devon. (1946.)
Brown, Miss J. E., 4 Fairfield Road, Woodford Green, Essex. (1948.)
Brown, Miss M. E., B.Sc., 11 Royal Gardens, Boston Road, Hanwell, London, W. 7. (1953.)
Brunner, Miss N., 9 Upper Clanbrasil Street, Dublin, Eire. (1949.)
Callan, I. W. McC., B.Sc., Dip.Agr.Sc., 19 Alexandra Road, Reading. (1950.)
Charlton, J. L., B.Sc., Dip.Bact., Imperial Chemical Industries Ltd., Hawthorndale Laboratories, Jealott's Hill Research Station, Bracknell, Berks. (1947.)
Clements, Miss C. A., B.Sc., Forest Products Research Laboratory, Princes Risborough, Aylesbury, Bucks. (1952.)
Cook, Mrs M. E. Ivimey, Westra Lodge, Dinas Powis, S. Wales. (1949.)
Dobbs, Mrs C. G., 101 Gladstone Terrace, Bangor, Caern. (1947.)
Elias, R. S., B.Sc., A.R.C.S., 22 Rythe Road, Claygate, Surrey. (1952.)
Ellis, Mrs M. B., 20 Palewell Park, East Sheen, London, S.W. 14. (1947.)
Geoghegan, Miss M. E., St Patrick's Hall, Reading. (1950.)
Gobbett, R. H., 11 Rostrevor Road, Fulham, London, S.W. 6. (1948.)
Goodhue, R. D., M.S.R., 34 Marshall Road, Rainham Mark, Gillingham, Kent. (1953.)
Gray, Miss M. R., 43 Woodcroft Avenue, Mill Hill, London, N.W. 7. (1947.)
Gregory, Mrs M. F., 47 Milton Road, Harpenden, Herts. (1945.)
Harper, W. O., 39 St James' Road, Watford, Herts. (1951.)
Hilhouse, Miss Agnes M., 6 Cornwall Road, Uxbridge, Middx. (1953.)
Howarth, Miss S., Royal Holloway College, Englefield Green, Surrey. (1951.)
James, L., 19 Bushey Road, Ickenham, Uxbridge, Middx. (1947.)
Johnson, Miss S. M.
Jones, Miss P. R., 2 Birchwood Mansions, Muswell Hill, London, N. 10. (1950.)
Jupp, T. H., St Peter's Parsonage, 10 Primrose Gardens, Hampstead, London, N.W. 3. (1952.)
Kelsall, Mrs H. M., 21c Stanhope Road, Highgate, London, N. 6. (1947.)
Kelsall, R. K., 21c Stanhope Road, Highgate, London, N. 6. (1947.)
King, J. M. B., 23 Lyncroft Gardens, Ealing, London, W. 13. (1945.)
Lee-Whiting, Mrs B. B., Wembury, Marine View Crescent, Rhos-on-Sea, N. Wales. (1950.)
Lock, Miss H. M., 99 Thames Drive, Leigh-on-Sea, Essex. (1947.)
Madelin, M. F., 8 Shaftesbury Cottages, Fenelon Place, London, W. 14. (1950.)
Martin, Miss J., B.Sc., 9 Enmore Gardens, East Sheen, London, S.W. 14. (1947.)
Metcalfe, C. R., B.A., Ph.D., Jodrell Laboratory, Royal Botanic Gardens, Kew, Surrey. (1947.)
Molesworth, Miss B., Studley College, Studley, Warwickshire. (1945.)
Montgomery, W. E., Forensic Science Laboratory, Shakespeare Street, Nottingham. (1950.)
Moore, Mrs W. C., Vallis, Amenbury Lane, Harpenden, Herts. (1945.)
Neville, W. J. J., 10 Penmaen Road, Conway, N. Wales. (1949.)
Nye, A. L., Forebrae, Butterstone, By Dunkeld, Perthshire. (1949.)
Pennington, Mrs H., Woodthorne, Wolverhampton. (1952.)

Pollard, Mrs R. M., 13 Abercorn Place, London, N.W. 8. (1949.)
Prentice, Mrs M. M., 24 Wordsworth Road, Harpenden, Herts. (1945.)
Rapson, H. D., 307 Grenville House, Dolphin Square, London, S.W. 1.
Read, Miss L. M., Research Station, East Malling, nr. Maidstone, Kent. (1946.)
Stevenson, Miss H. C., 11 Fosse Way, Ealing, London, W. 13. (1947.)
Tipler, Mrs M. K., B.Sc., 2 Waterside Lodge, Raymead Road, Maidenhead, Berks. (1945.)
Webb, Miss M. B., 22 Bristol Road, Chippenham, Wilts. (1948.)
Weston, Mrs S. D., Howe Farm House, Huntingdon Road, Cambridge. (1948.)
Wheeler, B. E. J., Department of Botany, University of Bristol. (1949.)
Whitton, B. A., 91 The Quadrant, Wimbledon, London, S.W. 20. (1952.)
Yalden, Miss M. E., 4 Somerfield Terrace, London Road, Maidstone, Kent. (1948.)

Members are requested to notify the Secretary (J. Webster) *and Treasurer* (W. Buddin) *of any change of address*

The Society gratefully acknowledges financial assistance with the publication of the *Transactions*: from Dr A. J. Juniper; Birkbeck College Publications Fund; Crown Agents for the Colonies; and The Wellcome Research Laboratories towards the cost of extra Plates: from Messrs Imperial Chemical Industries Ltd.; Glaxo Laboratories Ltd., John & E. Sturge Limited; The British Drug Houses Ltd.; and Watney Combe Reid & Co. Limited towards the cost of production of this volume: and to the Royal Society for a grant from the Scientific Publications Grant-in-aid towards the previous volume.

INDEX

New names are indicated by an asterisk

Absidia cylindrospora, 309
  glauca, 309
*Acaulopage acanthospora var. magna, 356–357
  stenospora, 357–358
Acrostalagmus cinnabarinus, 217, 309
  gonioides, 359–360
Ainsworth, G. C. 'The new Code of Botanical Nomenclature', 273–277
Ainsworth, G. C., *see* Reviews
Air spora, 260–266, 375–393
Alternaria, 90, 375, 380
  solani, sporulation of, 349–355
Aponogeton, Physoderma on, 347–348
Apricot (*Prunus persica*) bacteriosis, 38–45
Aquatic chytrids, list of British, 278–303
Arachniotus candidus, 217
Archangeliella beccarii, 127
  malaiensis, 128
Arthrobotrys oligospora, 360
Ascochyta linicola, 61–73
  usitatissima, 61, 62
Aspergillus amstelodami, 309
  awarmori, 202
  fischeri, 59, 309
  foetidus, 202
  malignus, 59
  nidulans, 309
  niger, 160, 198–209, 234–334
  *quadricinctus, 57–60
  sydowi, 309
  terreus, 309
  versicolor, 309
  violaceo-fuscus, 202
Asterionella formosa, chytrid parasites, of, 14–20

Bacteria on chytrids, 34–35
Basidiospores of the air spora, 389
Bawden, F. C., *see* Reviews
Blackwell, Elizabeth M. 'Haustoria of *Phytophthora infestans* and some other species', 138–158
Bose, S. K. and Mehta, P. R. 'A new species of *Entomophthora* on beetles', 52–56
Botryosporium longibrachiatum, 309
  pulchrum, 309
Botrytis cinerea, 309, 391
Brahmina, Entomophthora on, 52–55
Brian, P. W., Hemming, H. G., Moffatt, J. S. and Unwin, C. H. 'Canescin, an antibiotic produced by *Penicillium canescens*', 243–247
Brierley, W. B., *see* Reviews
British Mycological Society (1952):
  Acknowledgements (1953), 412
  Annual General Meeting, 164
  Forays: French (autumn), 170; London and provincial, 176; Perranporth (spring), 166
  List of members (1953), 394
  Plant Pathology Field Day, 176
  Proceedings, 163
  Receipts and Payments, 165
Brown, W., *see* Reviews
Burges, A. and Fenton, Enid. 'The effect of carbon dioxide on the growth of soil fungi', 104–108
Buxton, E. W. '*Schizonella melanogramma*' a new record for Britain', 196–197

Caeoma saxifragarum, 315
Canescin, 243–247
Canter, Hilda M. 'Annotated list of British aquatic chytrids', 278–303
Canter, Hilda M. and Lund, J. W. G. 'Studies on plankton parasites. II. The parasitism of diatoms with special reference to lakes in the English Lake District', 13–39
Cephalosporium acremonium, 309
  roseum, 309
  spp., 217, 309
Cercosporella herpotrichoides, 46–51
Cerebella, 75
Chaetomium cochloides, 309
  spinosum, 309
Chaetothyrium babingtonii, 162
*Chamonixia mucosa, 131
  *octorugosa, 133
Charlton, K. M. 'The sporulation of *Alternaria solani* in culture', 349–355
Chytridium versatile, 15, 20, 23, 27, 28
Chytrids, list of British aquatic, 278–303
Chytriomyces sp., 15
  tabellariae, 22
Cladosporium herbarum, 217, 260–266, 309, 375, 380–390
Claviceps deliquescens, 74
  lutea, 74
  paspali, 74–75
Coemansia pectinata, 309
Colletotrichum atramentarium, 340
  biologicum, 340
  fuscum, 335–343
  wahlenbergiae, 340
Coniothyrium sp., 217
Corner, E. J. H. and Hawker, Lilian E. 'Hypogeous fungi from Malaya', 125–137
Corticium coronilla, 307, 313
Crosse, J. E. 'Bacterial diseases of stone fruit trees in Britain. IX. Bacteriosis of apricot', 38–45
Cryptosporella eugeniae, 232
Cyclotella, chytrid parasites of, 31–32
Cylindrocarpon radicicola, 309

Dasyscypha calycina, 364
  willkommii, 367
  willkommii var. fuckelii, 365

*Dendrogaster aurantius, 134
  cambodgensis, 134
Dennis, R. W. G. and Ellis, M. B. 'Note: *Chaetothyrium babingtonii* (Berk.) Keissler', 162
Digitalis diseases, 335–346

*Elaphomyces carbonaceus, 137
  *singaporensis, 136
Elasmomyces borneensis, 129
  *malaiensis, 129
Ellis, M. B., *see* Dennis, R. W. G.
Emmons, C. W., *see* Reviews
*Entomophthora brahminae, 52–55
Erysiphe, 90, 375, 380

Fenton, Enid, *see* Burges, A.
Flax (*Linum*) foot rot, 61–73
Forays, *see* British Mycological Society
Fragilaria crotonensis, chytrid parasites of, 20–25
Fusarium culmorum, 200, 309
  equiseti, 200
  oxysporum, 200, 309
  solani, 200

Geomyces cretaceus, 309
Gibson, I. A. S. 'Crown rot, a seedling disease of groundnuts caused by *Aspergillus niger*', 198–209
Gibson, I. A. S. 'Crown rot, a seedling disease of groundnuts caused by *Aspergillus niger*. II. An anomalous effect of organo-mercurial seed dressings', 324–334
Gliocladium atrum, 217
  catenulatum, 310
  roseum, 217, 310
Gliomastrix convoluta, 104–107, 217
  convoluta var. felina, 105, 310
Gloeosporium digitalis, 340
Glynne, Mary D. 'Production of spores by *Cercosporella* herpotrichoides', 46–51
Gregory, P. H., *see* Reviews
Groundnut (*Arachis*) crown rot, 198–209
  control of, 324–334

Hawker, Lilian E., *see* Corner, E. J. H.; Reviews
Helicodendron giganteum, 114
  *multicatenulatum, 115
  paradoxum, 113
  triglitziensis, 114
  tubulosum, 114
  westerdijkae, 117
Helicoon fuscosporum, 112
Hemming, H. G., *see* Brian, P. W.
Henderson, D. M. 'Some Scottish rust fungi', 315–319
Hickman, C. J., *see* Reviews
Hirst, J. M. 'Changes in atmospheric spore content: diurnal periodicity and the effects of weather', 375–393
Hughes, S. J. '*Phragmocephala* in pure culture', 210–214
Hyde, H. A. and Williams, D. A. 'The incidence of *Cladosporium herbarum* in the outdoor air at Cardiff 1945–50', 260–266
*Hydnangium echinulatum, 126
  *virescens, 125
Hymenogaster mucosus, 131
  viscidus, 135
Hypogeous fungi from Malaya, 125–137
*Hysterangium purpureum var. malaiense, 135

International Code of Botanical Nomenclature, 273–277
Isaac, I. 'A further comparative study of pathogenic isolates of *Verticillium V. nubilum* Pethybr. and *V. tricorpus* sp. nov.', 180–195

Juniper, A. J. 'Some predacious fungi occurring on dung', 356–361

Kerr, A. 'Foot rot of flax caused by *Aschochyta linicola*', 61–73

Lachnea calycina, 364
Lachnella calycina, 364
  hahniana, 364
Lagenidium cyclotellae, 32
Lambertella corni-maris, antibiotics of, 109–110
Langdon, R. F. N. 'The genus *Ustilagopsis* Speg.', 74–75
Larch (*Larix*) canker, 362–373
Lund, J. W. G., *see* Canter, Hilda M.

Manners, J. G. 'Studies on larch canker. I. The taxonomy and biology of *Trichoscyphella willkommii* (Hart.) Nannf. and related species', 362–374
Mehta, P. R., *see* Bose, S. K.
Melampsora alpina, 315, 317
  arctica, 315, 317
  epitea, 315, 319
  epitea var. reticulata, 315–319
Melosira italica, chytrid parasites of, 25–27
*Micromycopsis oedogonii, 320–323
Moffatt, J. S., *see* Brian, P. W.
Mollison, Janet E. 'Effect of partial soil sterilization and acidification of soil on fungal population', 215–228
Monilia geophila, 217
Moore, W. C., *see* Obituary Notice
Mortierella hygrophila and var. minuta, 217
  isabellina, 309
  minutissima, 309
Mould growth, measurement of, 159–162
Mucor hiemalis, 217
  ramannianus, 105, 217, 309
  silvaticus, 309
  spinosus, 309

Nutman, F. J. and Roberts, F. M. 'Two new species of fungi on clove trees in Zanzibar Protectorate', 229, 234

Obituary Notice:
  Brooks, Prof. F. T. (by W. C. Moore), 177
Oedogonium, Micromycopsis on, 320–323
Oidiodendron griseum, 217

Paecilomyces spp., 217, 310
  varioti, 3

Papularia arundinis, 310
Pearson, A. A. 'Presidential Address: Homage aux mycologues français', 1–12
Pearson, A. A., *see* Reviews
Penicillium adamatzi, 217
  albidum, 310
  claviforme, 310
  expansum, 310
  canescens, 243–247
  frequentans, 161, 310
  funiculosum, 310
  janthinellum, 104, 217
  lilacinum, 217
  nigricans, 104–107, 217, 310
  phoeniceum, 310
  restrictum, 104, 217
  rugulosum, 105
  spinulosum, 104, 217, 310
  thomii, 310
  variable, 310
  vermiculatum, 217
  wortmanni, 160, 309
Perisporium vulgare, 309
Pestalotia adusta, 82
  neglecta, 83
  (*see also* Pestalotiopsis)
*Pestalotiopsis adusta, 82, 236
  *angusta, 241
  aquatica, 242
  dichaeta, 235
  disseminata, 84, 236
  funerioides, 87
  *gigas, 86
  glandicola, 86
  guepini and var. *major, 237
  *hughesii, 237
  maculiformans, 240
  mangiferae, 83, 239
  *natrassii, 82
  *neglecta, 83, 236
  *owenii, 88
  *owenii var. major, 89
  palmarum, 239
  papposa, 85
  royenae, 85
  theae var. minor, 85, 240
  versicolor, 86
  virgatula, 85
Phoma eupyrena, 217
  exigua, 61, 62, 68–69
  herbarum, 61–62, 69
  lini, 61, 62, 68–69
  linicola, 61, 63, 68
  sp. 61, 63, 69–70
Phragmocephala atra, 210–214
  cookei, 210–214
  minima, 214
  setosa, 214
Phyllosticta digitalis, 344–345
Physoderma aponogetonis, 347–348
Phytophthora, haustoria of, 138–158
Phytophthora cactorum, 154, 248–254
  cinnamomi, 255–259
  cryptogea, 255–259
  infestans, 138, 375, 280–390
Plankton parasites, 13
Plant pathological terms, 267
Polythrincium trifolii, 375, 380, 389
Pseudomonas cerasi, 38
  mors-prunorum, 38–45
  prunicola, 39, 41, 42, 44
  syringae, 39
Pyrenochaeta sp., 217
Pyronema confluens, 309
Reviews:
  Ainsworth, G. C., Medical mycology. An introduction to its problems, 1952 (by C. W. Emmons), 272
  Alexopoulos, C. J., Introductory mycology, 1952 (by G. C. Ainsworth), 270
  Le Bactériophage, 1953 (by F. C. Bawden), 271
  Barton-Wright, E. C. The microbiological assay of the vitamin B complex and amino acids, 1952 (by Lilian E. Hawker), 78
  Brooks, F. T. Plant diseases, Ed. 2, 1953 (by W. Brown), 268
  Index of fungi 1929, 1930, 1936–1939 (by G. C. Ainsworth), 77
  Josserand, M., La description des champignons supérieurs, 1952 (by A. A. Pearson), 271
  Konrad, P. and Maublanc, A. Les Agricales, Tome, 2, 1952 (by E. M. Wakefield), 270
  Langeron, M., Précis de mycologie, Ed. 2, 1952 (by G. C. Ainsworth), 77
  McKay, R. Sugar beet disease in Ireland, 1952 (by F. C. Bawden), 80
  Moreau, F., Les Champignons, Tome 1, 1953 (by C. J. Hickman), 269
  Mushroom Research Station reports, 1946–1951 (by P. H. G.), 80
  Pilát, A., KLIC Agricalium Europaeorum clavis dichotomica, 1951 (by A. A. P.), 271
  Plant Pathology (by W. B. Brierley), 79
  Roger, L., Phytopathologie des pays chauds, Tome 1, 1951 (by P. O. Wiehe), 78
Rhizophidium fragilariae, 20, 22, 23
  ? fusus, 27
  planktonicum, 14, 18, 19
Rhizopus arrhizus, 200
  stolonifer, 217, 309
Rhizosolenia, chytrid parasites of, 31
Roberts, F. M., *see* Nutman, F. J.
Roberts, Marjorie. '*Micromycopsis oedogonii* sp.nov', 320–323

Saxifraga rusts, 315–319
Schizonella melanogramma, 196–197
Sclerotium compactiusculum, 74–75
Scopulariopsis brevicaule, 310
Sepedonium sepedonioides, 310
Septoria digitalis, 343–344
Septosperma anomalum, 17, 22
  sp., 27
Soil fungi, 217, 309
  effect of carbon dioxide on, 104–108
Sparrow, F. K. 'Observations on chytridiaceous parasites of phanerogams. IV. *Physoderma aponogetonis* sp.nov. parasitic on *Aponogeton*, 347–348
Spicaria violacea, 104, 310

Spilsbury, J. 'Some fungus diseases of *Digitalis lanata*', 335–346
*Spirosphaera floriforme, 121
Sporotrichum spp., 310
Stachybotrys atra, 217
Stamps, D. Jean. 'Variation in a strain of *Phytophthora cactorum*', 248–259
Stamps, D. Jean. 'Oospore production in paired cultures of *Phytophthora* species', 255–259
Stanton, W. R. 'Field variation in *Venturia pirina*', 90–103
Stenton, H. 'The soil fungi of Wicken Fen', 304–314
Stephanodiscus, chytrid parasites of, 32–33
Stephanospora pengangensis, 130
Steyaert, R. L. 'New and old species of *Pestalotiopsis*', 81–89
Steyaert, R. L. '*Pestalotiopsis* from the Gold Coast and Togoland', 235–242
Stylopage lepte, 359
  rhynchospora, 358–359
Stysanus stemonitis, 310
Syncephalis, 309
Synedra, chytrid parasites of, 33–34

Tabellaria, chytrid parasites of, 27–31
Thiram as a groundnut seed-dressing, 324
Trichocladium asperum, 217
Trichoderma viride, 105, 215–228, 310
*Trichoscyphella hahniana, 364
  willkommii, 367
Trichothecium roseum, 217
Turner, J. N. 'Measurement of mould growth on solid substrates', 159–161

Unwin, C. H., *see* Brian, P. W.
Ustilago avenae, 379, 380–390
Ustilagopsis, 74
  bertoniensis, 75
  compactiuscula, 74
  delequescens, 74

*Valsa eugeniae, 229
Van Beverwijk, Agathe L. 'Helicosporous Hyphomycetes', 111–124
Venturia inaequalis, 380
  pirina, 90–130
Vermicularia herbarum, 340
Verticillium albo-atrum, 180–195
  dahliae, 180–195
  nigrescens, 180–195
  nubilum, 180–195
  *tricorpus, 180–195
Volutella ciliata, 310

Wakefield, E. M., *see* Reviews
*Wakefieldia, 130
  *striaespora, 130
Wicken Fen soil fungi, 304–314
Wiehe, P. O., *see* Reviews
Williams, D. A., *see* Hyde, H. A.
Wood, R. K. S. 'The antagonism of *Lambertella corni-maris* to fungi and bacteria, 109–110

Xanthomonas pruni, 38

Yuill, E. 'A new ascosporic species of *Aspergillus*', 57–60

Zygorhizium melosirae, 25, 34
  *planktonicum, 13–15, 33–35
  sp., 32
Zygorrhynchus vuillemini, 104–107

THE BRITISH MYCOLOGICAL SOCIETY TRANSACTIONS

Vol.	PART 1	2	3	4	5
1	*m*	*m*	6*s.*	*m*	*m*
2	[		68*s.*		]
3	[		80*s.*		]
4	*m*	*m*	—	—	—
5	*m*	*m*	*m*	—	—
6	*m*	*m*	(20*s.*	)	—
7	(	40*s.*		)	—
8	(	40*s.*		)	—
9	(20*s.*	)	*m*	*m*	—
10	(	40*s.*		)	—
11	[	40*s.*		]	—
12	(10*s.*)	*m*	*m*	(10*s.*)	—
13	*m*	*m*	*m*	*m*	—
14	[	40*s.*		]	—
15	*m*	*m*	*m*	*m*	—
16	*m*	[	30*s.*	]	
17	*m*	*m*	[10*s.*]	*m*	—
18	*m*	[	30*s.*	]	—
19	*m*	(	30*s.*	)	—
20	(	40*s.*		)	—

Vol.	PART 1	2	3	4
21	(		40*s.*	)
22	(		40*s.*	)
23	(		40*s.*	)
24	(		40*s.*	)
25	(		40*s.*	)
26	(		40*s.*	)
27	(		40*s.*	)
28	(		40*s.*	)
29	(		40*s.*	)
30	(	Jubilee Vol. 20*s.*		)
31	[25*s.*	]	*m*	*m*
32	[25*s.*	]	*m*	*m*
33	(		50*s.*	)
34	(		50*s.*	)
35	(		50*s.*	)

m = out of print, available on microfilm to order
( ) = volume (or part volume) sold separately
[ ] = only sold as part of a long set of volumes

REPRINTS AND MISCELLANEOUS PUBLICATIONS

PLANT PATHOLOGY COMMITTEE: *List of Common British Plant Diseases*, 3rd Ed. 1945. Price net, 5*s.* Postage, 4*d.*

A. A. PEARSON: *Cooke's illustrations of British Fungi* (*Annotations by L. Quélet, R. Maire & C. Rea*). *Ex* Vol. 20, Pt. 1. 1935. Price net, 2*s.* 6*d.* Postage, 3*d.*

T. PETCH: *British Hypocreales*. *Ex* Vol. 21, Pts. 3 & 4. 1938. Price net, 2*s.* Postage, 3*d.*

A. A. PEARSON & R. W. G. DENNIS: *Revised list of British Agarics and Boleti*. *Ex* Vol. 31, Pts. 3 & 4. 1948. Price net, 5*s.* Postage, 3*d.*

J. RAMSBOTTOM & F. L. BALFOUR-BROWNE: *List of Discomycetes recorded from the British Isles*. *Ex* Vol. 34, Pt. 1. 1951. Price net, 5*s.* Postage, 3*d.*

FIFTY YEAR INDEX TO THE TRANSACTIONS, Vols. 1–30; 1896–1946. Compiled by G. R. BISBY. Price net, 21*s.* Postage, 6*d.*

Enquiries and orders for the above to be addressed to

CAMBRIDGE UNIVERSITY PRESS
200 EUSTON ROAD, LONDON, N.W. 1
or to
CAMBRIDGE UNIVERSITY PRESS
AMERICAN BRANCH, 32 EAST 57TH STREET, NEW YORK 22

FORAY COMMITTEE PUBLICATIONS

(*To be obtained from the Treasurer and on sale at meetings and forays*)

A key to the genera of the agarics and Boleti. By A. A. PEARSON. 32 pp. 1950. Price net, 2*s.* 6*d.* Postage, 2*d.*

Guide to the literature for the identification of British fungi. 1952. Price net, 2*s.* 6*d.* Postage, 2*d.*

CONTENTS

PAGE

The new Code of botanical nomenclature. By G. C. Ainsworth 273

Annotated list of British aquatic chytrids. By Hilda M. Canter 278

The soil fungi of Wicken Fen. By H. Stenton. (With 3 Text-figures) 304

Some Scottish mountain rust fungi. By D. M. Henderson 315

Micromycopsis oedogonii sp. nov. By Marjorie Roberts. (With Plate 17 and 12 Text-figures) 320

Crown rot, a seedling disease of groundnuts caused by *Aspergillus niger*. II. An anomalous effect of organo-mercurial seed dressings. By I. A. S. Gibson 324

Some fungus diseases of *Digitalis lanata*. By J. F. Spilsbury. (With Plates 18–20 and 5 Text-figures) 335

Observations on chytridiaceous parasites of phanerogams. IV. *Physoderma aponogetonis* sp.nov. parasitic on *Aponogeton*. By F. K. Sparrow. (With 1 Text-figure) . . 347

The sporulation of *Alternaria solani* in culture. By K. M. Charlton. (With Plate 21) . 349

Some predacious fungi occurring in dung. By A. J. Juniper. (With Plate 22 and 4 Text-figures) 356

Studies on larch canker. I. The taxonomy and biology of *Trichoscyphella willkommii* (Hart.) Nannf. and related species. By J. G. Manners. (With Plate 23 and 6 Text-figures) 362

Changes in atmospheric spore content: diurnal periodicity and the effects of weather. By J. M. Hirst. (With Plate 24 and 5 Text-figures) 375

List of Members 394

Acknowledgements 412

Index 413

INSTRUCTIONS TO AUTHORS

Contributors are asked to be clear and concise, and to use a recent issue of the *Transactions* as a guide to the arrangement of headings and subheadings, explanations of plates and figures, references to the literature, and other details. Each paper should start with a short summary.

The standards adopted in the *Transactions* are those outlined in the Royal Society's pamphlet, *General Notes on the Preparation of Scientific Papers* (1950, Cambridge University Press, 200 Euston Road, London N.W. 1, price 2*s*. 6*d*.). An exception is that in the *Transactions* citations of references to literature, although based on the Harvard system, include the full titles of the papers and complete page numbers. Titles of journals are abbreviated in accordance with *A World List of Scientific Periodicals*, 1952.

The scientific names of genera and species are printed in italics and should be underlined in the typescript. The title of the paper, headings, and subheadings may be in capitals, but should not be underlined.

The attention of authors proposing new taxonomic groups is drawn to the need to give an adequate description and figure, and to designate the types as required by the *International Code of Botanical Nomenclature*. The description of a new species should be accompanied by a figure, and whenever possible the type specimen should be deposited in one of the national herbaria.

The British Mycological Society subscribes to the Fair Copying Declaration, details of which may be obtained from the offices of the Royal Society.

Printed in Great Britain at the University Press, Cambridge (*Brooke Crutchley, University Printer*)